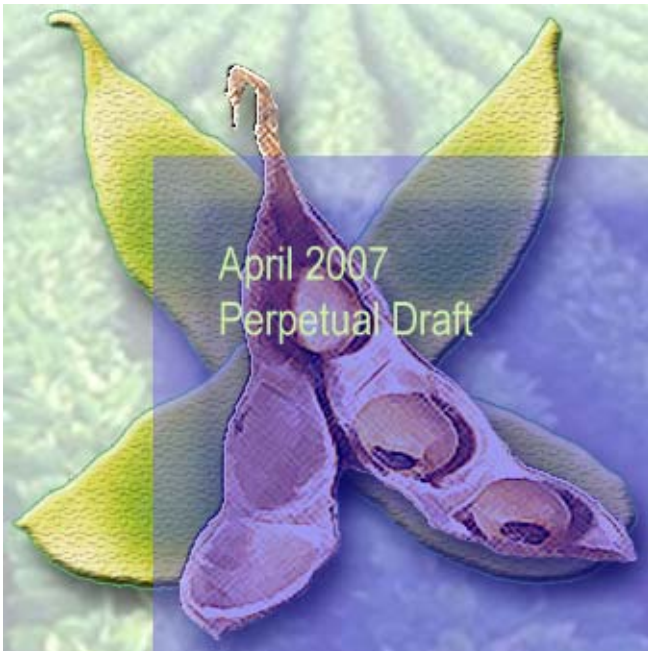

April 2007
Perpetual Draft

soybean commodity based survey

caps

cooperative agriculture pest survey

TABLE OF CONTENTS

AUTHORS, REVIEWERS, DRAFT LOG 5

INTRODUCTION TO REFERENCE 6

SOYBEAN BACKGROUND 11

SOYBEAN PESTS 14

Arthropod Pests 14
- **Aphids** 14
  - ***Aphis glycines*** 14
  - ***Aulacorthum solani*** 19
- **Beetles/Weevils** 25
  - ***Adoretus sinicus*** 25
  - ***Diabrotica speciosa*** 34
  - ***Epilachna varivestis*** 39
  - ***Naupactus xanthographus*** 44
  - ***Popillia japonica*** 48
- **Flies** 54
  - ***Melanagromyza dolichostigma*** 54
  - ***Melanagromyza sojae*** 59
  - ***Ophiomyia centrosematis*** 64
  - ***Ophiomyia phaseoli*** 68
  - ***Rivellia quadrifasciata*** 74
- **Mealybugs** 77
  - ***Maconellicoccus hirsutus*** 77
- **Mites** 84
  - ***Eutetranychus africanus*** 84
  - ***Eutetranychus orientalis*** 87
  - ***Tetranychus kanzawai*** 90
- **Moths** 92
  - ***Anticarsia irrorata*** 92
  - ***Autographa gamma*** 94
  - ***Chrysodeixis chalcites*** 100
  - ***Crocidosema aporema*** 105
  - ***Cydia fabivora*** 108
  - ***Etiella zinckenella*** 112
  - ***Helicoverpa armigera*** 117
  - ***Lampides boeticus*** 123
  - ***Leguminivoria glycinivorella*** 128
  - ***Mamestra brassicae*** 132
  - ***Spodoptera littoralis*** 136
  - ***Spodoptera litura*** 141
- **Stink Bugs** 147
  - ***Acrosternum hilare*** 147
  - ***Nezara viridula*** 147
  - ***Euschistus servus*** 147
  - ***Halyomorpha halys*** 147

Thrips 150
Frankliniella intonsa 150
Thrips palmi 155

Diseases 163
Bacterial Diseases 163
Pseudomonas savastanoi pv. glycinea 163
Xanthomonas axonopodis pv. glycines 168
Fungal Diseases 173
Cercospora kikuchii 173
Colletotrichum lindemuthianum 178
Colletotrichum truncatum 185
Disported phaseolorum var. sojae 191
Fusarium graminearum 196
Passalora sojina 205
Peronospora manshurica 212
Phakopsora pachyrhizi 217
Septoria glycines 227
Viral Diseases 231
Bean Common Mosaic Virus (BCMV) 231
Bean Golden Mosaic Virus (BGMV) 237
Bean Pod Mottle Virus (BPMV) 247
Indonesian Soybean dwarf virus (ISDV) 251
Mung Bean Yellow Mosaic Virus (MYMV) 253
Peanut Stunt Virus (PSV) 257
Red Clover Mottle Virus (RCMV) 263
Soybean Dwarf virus (SbDV) 266
Soybean Mosaic Virus (SMV) 272

Other pests 280
Nematodes 280
Aphelenchoides besseyi 280
Ditylenchus africanus 286
Ditylenchus destructor 292
Heterodera glycines 298
Rotylenchulus macrodoratus 307
Rotylenchulus reniformis 310
Xiphinema ifacolum 317
Parasitic Plants/Weeds 320
Alectra vogelii 320
Borreria latifolia 324
Commelina benghalensis 327
Euphorbia heterophylla 332
Mimosa diplotricha 338
Pharbitis nil 343
Mollusks 346
Cernuella virgata 346

Glossary 350

Appendix A: Diagnostic Resource Contacts 378

Appendix B - References 380
APHIDS 380
BEETLES AND WEEVILS 382

FLIES ***388***
MEALYBUGS ***390***
MITES ***395***
MOTHS and BUTTERFLIES ***397***
STINKBUGS ***414***
THRIPS ***414***
BACTERIAL DISEASES ***419***
FUNGAL DISEASES ***422***
VIRAL DISEASES ***436***
NEMATODES ***452***
PARASITIC PLANTS AND WEEDS ***459***
MOLLUSKS ***474***

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Introduction to Reference

History of Commodity Based Survey

The CAPS community is made up of a large and varied group of individuals from federal, state, and university organizations who utilize federal (and other) funding sources to survey for, and (in some cases) diagnose exotic and invasive plant pests. By finding pests early, eradication efforts will likely be less expensive and more efficient. For more information on CAPS, access to the Program Guidebook may be found at the following URL.

http://www.aphis.usda.gov/ppq/ep/pestdetection/CAPSGuidebookComplete.PDF

Traditionally, states have been given a list of pests. Each year states choose (from this list) a number of pests to incorporate in their own specialized surveys. There is certainly value surveying for plant health threats in terms of discreet pests. However, this may not always be the most efficient means of survey. For example, a single pest may occur on a myriad of different hosts, making a comprehensive survey too time consuming and expensive. An alternative method has been suggested. Grouping important pests under the umbrella of a single commodity could be a more efficient way to look for certain pests. The rationale for choosing a commodity survey in certain instances includes the following:

Survey area will be smaller and targeted.

Resources can be better utilized with fewer trips to the field

Commodities are easy to prioritize in terms of economic and regional (geographic) importance.

The Center for Plant Health Science and Technology (CPHST) has been charged to develop a commodity-based survey strategy in support of the CAPS program. There are two types of end products being developed for each commodity. Each product serves a valuable yet very different purpose. The result is a set of paired documents developed for each commodity. A description of these documents is included below:

> **Commodity Based Survey Reference (CSR):** This document is comprised of a series of pest data sheets, mini-PRAs, or early detection PRAs. The data sheets are highly graphic and illustrate the biology, survey, and identification of particular pests in appropriate detail for CAPS surveyors. The pests in this document are numerous. The pests were chosen from a number of sources, including the CAPS National Pest list, Regional pest lists, the Global Pest and Disease Database, and the CABI Compendium. States are not required to survey for all of the pests in this document, but may choose those that are particularly relevant to include in

their survey. In general, this document should serve as a desk reference for survey specialists as they plan their cooperative agreements. It may also be useful to obtain high quality scientific information quickly within the field season.

Commodity Based Survey Guidelines (CSG): This document is smaller. The list of pests is shorter than those chosen for the CSR. A subcommittee of the CAPS National Committee determines which pests from the CSR will be included in the CSG; As such, states that participate in these surveys must survey for all organisms listed in the CSG. The CSG sets forth guidelines for survey and identification from a broad scale (site selection, number of acres to survey, number of samples to take, etc.) and a narrow scale (field methods, survey tools, transporting samples, etc.). States are encouraged to follow the procedure set forth in the CSG. The methods are intended to increase homogeneity of the national data set, and increase the statistical confidence in negative data (e.g. demonstration of "free from" status).

As a pilot project, Citrus was undertaken as the first commodity in this initiative. The products were developed for implementation in the 2007 survey. Citrus was chosen because it is an economically important commodity that is equally distributed in both regions. Moreover, it is also grown on relatively low acreage in few overall states. To date, survey strategies for pests of citrus are also well documented. Since then, several other commodities have been undertaken, including soybeans, cotton, small grains, and eastern forests.

Soybean Commodity Survey Reference

The *Soybean Commodity Survey Reference* (CSR) is a companion document to the *Soybean Commodity Survey Guidelines* (CSG). Both documents are intended to be tools to help survey professionals develop surveys for exotic soybean pests. The *Soybean CSR is* a collection of detailed datasheets on pests of soybean, including exotics and endemics. Additionally, the authors have tried to identify pests which may be easily confused with or potential vectors of exotic pests. These datasheets contain detailed information on the biology, host range, survey strategy, and identification of these pests.

In contrast, the *Soybean Commodity Survey Guideline* companion document is intended to help states focus resources on survey efforts and identification of a smaller group of target pests (usually less than a dozen). The guidelines contain little information about biology. Moreover, they focus on survey design, sampling strategies, and methods of identification. There is no silver bullet survey that would be wholly applicable to each location in the United States. Environment, personnel, budgets, and resources vary from state to state. Thus, the guidelines will provide a template that states can use to increase the uniformity and usability of data across political, geographic, and climatic regions, while maintaining flexibility for appropriateness within individual regions.

Purposes

To relate scientific information on a cadre of threatening pests.

To collect pest data at a sub-regional, regional, and national level versus data collected from a single location.

To help develop yearly surveys.

To help CAPS cooperators increase their familiarity with exotic pests and commonly confused pests that are currently found in a given commodity.

To help with identification and screening of pests sampled from the field.

To collate a large amount of applicable information in a single location.

End Users

As previously noted, this document may be used for many purposes. Likewise, it will be of value to numerous end users. As the document was developed, the authors specifically targeted members of the CAPS community who are actively involved in the development and implementation of CAPS surveys.

- **State Plant Health Director** (SPHD): The SPHD is the responsible PPQ official who administers PPQ regulatory and pest detection activities in his or her State. The SPHD is also responsible for ensuring that the expanded role of CAPS is met in his or her State. In many States, the SPHD needs to provide guidance for the State's ongoing management of pest risk and pest detection. However, SPHD responsibilities will vary according to each State's ability to carry out the various components of the CAPS Program.
- **State Plant Regulatory Official** (SPRO): These individuals are employees of their respective states and generally manage the expanded survey program. The SPRO is the responsible State official who administers State agricultural regulatory programs and activities within his or her respective State.
- **Pest Survey Specialists** (PSS): The PSS, a PPQ employee, is supervised by the SPHD of the State in which he or she is assigned. A PSS may also be responsible for survey activities and work with the SSC and the Survey Committee in more than one State.
- **State Survey Coordinators** (SSC): The SSC is a State employee responsible for coordinating each State's CAPS Program, participating as a member of the SCC, and acting as liaison with the State PPQ office. Each core CAPS Program will develop a network with other agencies, nongovernmental organizations, and members of SCC's. This networking will ensure the coordinated use of existing State and national resources, such as dollars and in kind contributions, in the evaluation of risks of specific exotic plant pests and weeds. State survey priorities will be set accordingly.

Diagnosticians: Diagnostic capabilities vary by state. Some states have advanced networks of diagnosticians, whereas other states access diagnostic support through National Identification Services (NIS) or through contracts with external partners. States are encouraged to utilize qualified diagnosticians in their respective states if expertise is available. PPQ offers diagnostic support for the CAPS program through NIS. A major responsibility for NIS's Domestic Identifiers is to provide diagnostic support to CAPS programs. There are plant pathology and entomology domestic identifiers in each of the regions. A Forest Entomology Domestic Identifier oversees both regions. To learn more about diagnostic resources available to you, discuss your diagnostic requirements and options with your State Plant Health Director, one of the regional Domestic Identifiers, and/or NIS. Appendix A has a listing of NIS and Domestic Identifier contact information.

Organisms Included in the Soybean Survey Reference

Organisms are organized first by pest type, *e.g.* arthropods, diseases, weeds, nematodes, and mollusks. Next, organisms are divided by their broad taxonomic classification. For example, the pest type "arthropods" contains groups of flies, moths, aphids, *etc.* Next, organisms are arranged alphabetically by their scientific names. Common names are included as well.

As previously mentioned, organisms were chosen for the reference from a variety of sources, including the CAPS National List, the Eastern & Western Region Pest Lists, the Global Pest and Disease Database, CABI Crop Compendium, and others. Pests may be exotic, regionally established or endemic. Some pests were chosen because they can be confused with target pests. Still, others were chosen because they vector a pest of concern. There is no obligation to survey for all pests listed in this reference. Moreover, the reference is intended to empower states to choose which pests might be appropriate targets within their borders.

To help provide a rationale for the inclusion of each pest in the reference, the authors have included a symbol in the table of contents and upper right hand of pest introduction pages. These symbols indicate that the pest is a CAPS Target, an Eastern Regional Threat, a Western Regional Threat, an Emerging Pest, an Endemic Pest, a Commonly Confused Pest, a Vector, or a National Threat. An explanation of each of these symbols follows.

CAPS Targets are listed on the CAPS National Pest List. These are organisms that have been through a rigorous prioritization process, and have been determined to pose a significant threat to the United States.

Eastern Regional Threats are listed on the ER Pest List. Some of these organisms may be regionally established in the United States. However, the ER has determined that they pose a particular risk for new establishments or further spread.

Western Regional Threats are listed on the WR Pest List. Some of these organisms may be regionally established in the United States. However, the WR has determined that they pose a particular risk for new establishments or further spread.

Emerging Pests are listed on the Emerging Pest List, but not on the ER or WR Pest List. These organisms are regionally established and may have the potential to become larger problems as they spread.

Endemic Pests are widely established in the United States. These pests are included in the reference to help surveyors develop a familiarity for what they are "likely" to encounter when they are conducting field surveys.

Commonly Confused Pests are endemic pests that may resemble the pest of concern (as with arthropods), or may induce symptoms that resemble symptoms caused by pests of concern (as with pathogens).

Vectors may be exotic of endemic organisms. Vectors have an association with a pest of concern and often serve as an avenue for dissemination for such organisms.

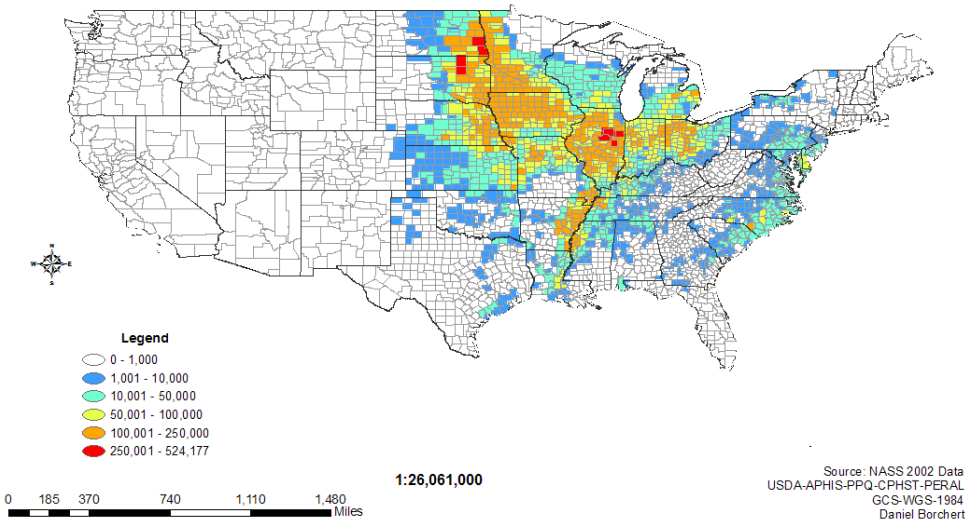
National Threats are pests that have been cited (in the GPDD, CABI Crop Protection Compendium, Homeland Security, and Select Agent lists) as **not known to be present** in the United States. National Threats are not associated with CAPS Targets, and are not present on the CAPS National List.

Soybean Background

Soybean is a member of the family Leguminoseae, subfamily Papilionaceae. It is an annual, erect bushy plant. The flowers are borne on short axillary or terminal racemes. The flowers are normally self-pollinated and completely self-fertile. Soybean is mainly grown in areas where the summer is hot and humid; however, it does withstand extreme summer and winter temperatures. The optimum temperature for growing soybean is 25 to 30 °C. Well-drained sandy or clay loams and alluviums with good fertility are generally suitable for the cultivation of the crop.

For many years, soybean acreage increased very slowly. There were only 1.8 million acres in the United States in 1924 when the first official estimates became available. At that time, most of the crop was used for hay. Following World War II, soybean production moved from the southern U.S. into the Corn Belt. The major soybean producing states of Iowa, Illinois, Minnesota, Indiana, Ohio, Missouri, and Nebraska produced 67 percent of the U.S. total in 2003; the southern and southeastern states of Arkansas, Mississippi, North Carolina, Kentucky, Tennessee, Louisiana, Alabama, and Georgia produced 14 percent. Other states with significant soybean acreage are South Dakota, Kansas, Michigan, Wisconsin, and North Dakota. The USDA estimates the 2005 U.S. soybean acreage at 73.0 million acres.

Soybean Acreage by County
2002 NASS data



Vegetative Stages. Vegetative stages are determined by counting the number of nodes on the main stem, beginning with the unifoliolate node, which have or have had a completely unrolled leaf (Fehr et al., 1971). The unifoliate node is the first node on a plant where true leaves develop. A leaf is considered completely unrolled when the leaf at the node immediately above it has unrolled sufficiently so the two edges of each leaflet are no longer touching. At the terminal node on the main stem, the leaf is considered completely unrolled when the leaflets are flat and similar to appearance to older leaves on the plant. Description of vegetative stages is given in Table 1.

Table 1. Vegetative stages and developmental descriptions of soybean.

Stage no.	Description
V1	Completely unrolled leaf at the unifoliolate node.
V2	Completely unrolled leaf at the first node above the unifoliolate node.
V3	Three nodes on the main steam beginning with the unifoliolate node.
V (N)	N nodes on the main stem beginning with the unifoliolate node.

Reproductive Stages. Reproductive stages are determined by examining the flowers and pods at the upper portion of the main stem, which is suitable for genotypes in all environments (Fehr et al., 1971). Description of reproductive stages is given in Table 2.

Table 2. Reproductive stages and developmental descriptions of soybean.

Stage no.	Description
R1	One flower at any node.
R2	Flower at node immediately below the uppermost node with a completely unrolled leaf.
R3	Pod 0.5 cm (1/4 inch) long at one of the four uppermost nodes with a completely unrolled leaf.
R4	Pod 2 cm (3/4 inch) long at one of the four uppermost nodes with a completely unrolled leaf.
R5	Beans beginning to develop (can be felt when the pod is squeezed) at one of the four uppermost nodes with a completely unrolled leaf.
R6	Pod containing full size green beans at one of the four uppermost nodes with a completely unrolled leaf.
R7	Pods yellowing; 50% of leaves yellow. Physiological maturity.
R8	95% of pods brown. Harvest maturity.

References:

Fehr, W.R., Caviness, C.E., Burmond, D.T., and Pennington, J.S. 1971. Stage development descriptions for soybeans, *Glycine max* (L.) Merrill. Crop Science 11:929-931.

Gibson, L. and Benson, G. 2005. Origin, History, and Uses of Soybean (*Glycine max*). Iowa State University. http://www.agron.iastate.edu/courses/agron212/Readings/Soy_history.htm

Soybean Pests

Arthropod Pests

Aphids

Aphis glycines

Scientific Name
Aphis glycines Matsumura

Synonyms:
Aphis justiceae

Common Name(s)
Soybean aphid

Type of Pest
Aphid

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Aphididae

Reason for Inclusion in Manual

Pest Description
Aphis glycines is a small yellow aphid with black siphunculi (cornicles) (Blackman and Eastop, 1984) (Fig. 1). Takahashi et al. (1993) presented biometric data, including body sizes: 1.89 mm for virginoparous aptera, 1.75 mm for virginoparous alata, 2.02 mm for gynopara, 1.5 mm for ovipara, 1.68 mm for alate males, and 1.87 mm for both fundatrix and apterous fundatrigenia.

Biology and Ecology:
A. glycines may have 15 to 18 generations a year, but must have access to buckthorn (*Rhamnus* spp.) to produce eggs for overwintering. In the spring, two

wingless generations develop on buckthorn before the first winged migrants are produced that re-infest soybean (CABI, 2004).

Pest Importance

A. glycines is distributed throughout the Far East, principally in China, Japan, Indonesia, Thailand, Korea, far eastern Russia, North Borneo, peninsular Malaysia and the Philippines (Blackman and Eastop, 2000). It is a major pest of soybean in China, causing particularly severe economic losses in the regions of Jilin, Liaoning, Heilongjiang and Neimenggu (Wang et al., 1962). *Aphis glycines* is capable of affecting both growth and seed production in soybeans. Economic infestations may occur from mid vegetative stages through pod fill. In China, Wang et al. (1996) found a 27.8% reduction in seed yields and a 20.2 cm decrease in plant height in infested plants compared with controls.

Figure 1. (Left) Wingless adult female. (Right) Winged adult female. *Aphis glycines,* is a small pale yellow aphid with black cornicles (siphunculi) and pale cauda. Its size and color distinguish it from other aphids on soybean. Photo courtesy of *1681H*http://www.ceris.purdue.edu/napis/pests/saphid/aglycin.html

It has recently been introduced into Australia, Canada and the U.S. (Fletcher and Desborough, 2000; Michelutti et al., 2002; Edwards et al., 2001). It has quickly become a soybean pest in North America, where aphids had previously not attacked soybeans (DiFonzo and Hines, 2001). *A. glycines* causes economic damage due to direct feeding and indirect damage due to the spread of viruses, in particular soybean mosaic virus (SMV).

A. glycines is a vector of a range of plant viral diseases, including soybean mosaic, soybean stunt (cucumber mosaic virus), abaca mosaic, beet mosaic, millet red leaf, mungbean mosaic, bean yellow mosaic and Indonesian soybean dwarf (Blackman and Eastop, 2000).

Symptoms/Signs

Infestation peaks of *A. glycines* during the bloom stage may cause stunted soybean plants, reduced pod and seed counts, and distorted leaves. Distorted, curled, yellowed, and wilted leaves can also be found on heavily infested plants later in the season. Honeydew and sooty mold on all plant stages are indicative

of *A. glycines* infestations. *Aphis glycines* populations can build at any time from early vegetative stages through bloom (Fig. 2). Initially, colonies establish on new leaves in the outer canopy, later penetrating deeper into the canopy and moving to the undersides of leaves as plants mature (CABI, 2004).

Direct damage by *A. glycines* is mainly due to feeding on young growing stems and leaves. Nutrients and water are taken directly from the plant's vascular system, through the aphid's stylet. Large infestations of aphids cause leaf distortion and loss of plant vigor, stunted plant growth, reduced pod and seed production, and subsequent yield losses (CABI, 2004).

Soybean infected with soybean mosaic virus has leaves with vein-clearing and chlorotic symptoms. Plants become stunted with shortened petioles and internode lengths, and defoliation may lead to plant death (Quimio and Calilung, 1993).

Figure 2. Large population of soybean aphid on soybean. Photo courtesy of Marlin Rice, Iowa State University. 1680Hhttp://www.ent.iastate.edu/imagegal/homoptera/aphid/soybeanaphid/soybean_aphids_boone.html

Known Hosts

The winter hosts of *A. glycines* are *Rhamnus* spp. (family Rhamnaceae), usually *Rhamnus davurica* in Asia. Several native and introduced species of *Rhamnus* (buckthorn) are common in North America, where *A. glycines* has recently been introduced, although only the exotic species *R. davurica* and *R. cathartica* have been confirmed as winter hosts (Voegtlin, 2002).

The summer host range is restricted to certain Fabaceae. In addition to cultivated soybean, it has been found on wild *Glycine* spp. (Wang et al., 1962) and has also been recorded from *Pueraria phaseoloides*, *Desmodium intortum* and a limited range of other wild hosts (Blackman and Eastop, 1984). It is the only aphid known to colonize soybeans.

Major hosts:
Glycine max (soybean)

Minor hosts:
Pueraria phaseoloides (tropical kudzu) and *Rhamnus davurica* (buckthorn).

Wild hosts:
Glycine spp.

Known Distribution

A. glycines is native to China and is widely distributed in soybean-growing regions of the Far East. It is common on soybean in China (Wang et al., 1962) and occurs in soybean fields in the Philippines (Quimio and Calilung, 1993), Japan (Takahashi et al., 1993), Indonesia (Iwaki, 1979), Thailand (Napompeth, 1978), North and South Korea, Malaysia and North Borneo (Blackman and Eastop, 1984). *A. glycines* has recently been found in Australia but only on soybeans (Fletcher and Desborough, 2000).

Potential Distribution Within the US

A. glycines was first observed in the U.S. in July 2000, in Wisconsin. It may have been present previously, but the conditions in 2000 were particularly favorable for aphids (University of Wisconsin, 2000). By 2002, it had infested soybeans in many states in the Midwest and North Central regions of the U.S., and also in Ontario, Canada (Edwards et al., 2001; Anon., 2002; Michelutti et al., 2002). It was found overwintering on *Rhamnus cathartica* in Iowa in the spring of 2002 (Anon., 2002).

Survey

To survey for soybean aphid, look for colonies of *A. glycines* in stem apices and young leaves of the growing soybean plants (Fig. 2, 3). On mature plants, colonies are also found on the underside of larger leaves.

Figure 3. Soybean aphid colony on soybean. Photo courtesy of *1679H*http://www.ceris.purdue.edu/napis/pests/saphid/aglycin.html

A. glycines can migrate from buckthorn to soybeans at different times in different regions depending upon environmental cues. Soybean fields should be monitored regularly as plants develop from the seedling stage (V2) through bloom (R1 to R2), because plants remain suitable for *A. glycines* throughout this period. In addition, *A. glycines* distribution across plant parts changes with plant development. Although leaves are most convenient to sample, the only reliable sampling unit is the entire plant.

Key Diagnostics

Takahashi et al. (1993) described the characteristic features of *A. glycines*, which could be used to separate it from other species (e.g. the number of secondary rhinaria, the number of caudal setae, the length of siphunculi, and the length of the last rostral segment). On legumes, *A. gossypii* has a shorter, darker cauda than *A. glycines*. *A. craccivora* has a black patch on the back of wingless adults, that is absent in *A. glycines*. Adult *A. woglumi* have metallic grey wings with light markings, red abdomens, white-yellow legs and antennae (CABI, 2004).

When it was first observed in the U.S., introduced *A. glycines* had to be distinguished from the morphologically similar *A. gossypii* using a high-powered microscope. Now that *A. glycines* is established, it can be assumed that aphid colonies on soybeans in North America are *A. glycines*, as no other species is known to develop colonies on soybeans in the U.S. (Voegtlin, 2002).

Aulacorthum solani

Scientific Name
Aulacorthum solani Kaltenbach (Asian strain)

Synonyms:
Acyrthosiphon solani, Aphis solani, Aulacorthum methae, Dysaulacorthum pseudosolani, Dysaulacorthum vincae, Macrosiphum aucubae, Macrosiphum piceaella, Macrosiphum pseudosolani, Macrosiphum vincae, Myzus aquilegiae, Myzus chelidonii, Myzus duffieldi, Myzus gei, Myzus glaucii, Myzus hydrocotylae, Myzus mercurialis, Myzus neogei, Myzus piceaellus, Myzus polyanthi, Myzus pseudosolani, Myzus solani, Phorodon solani, Rhopalosiphum solani, Siphoniella solani, Siphonophora solani, Macrosiphum solani, Aulacorthum pseudosolani, Aphis solani, Aulacorthum circumflexum, Macrosiphum matsumuraesanum, Myzus lamii

Common Name(s)
Foxglove aphid, greenhouse potato aphid, green potato aphid, dock aphid, potato aphid

Type of Pest
Aphid

Taxonomic Position
Class: Insecta (Hexapoda), **Order:** Hemiptera, **Family:** Aphididae

Reason for Inclusion in Manual

Figure 1. Apterous adults, nymphs, and winged adult on petiole of potato. Photo courtesy of Robert Y., INRA Rennes 1677Hhttp://www.inra.fr/Internet/Produits/HYPPZ/RAVAGEUR/3aulsol.htm

Pest Description
Kaltenbach (1843) first described the foxglove aphid from potato and named it *Aphis solani*. Subsequently, other workers described this polyphagous species under numerous names. Hille Ris Lambers (1949) comprehensively reviewed the synonymy and clearly established the validity of *Aulacorthum solani* as the correct name of the foxglove aphid. Only two of the many names proposed for the aphid have been used

extensively. Theobald (1922) described the insect as *Myzus pseudo-solani*, which is now considered to be a synonym.

Nymph: Inner faces of frontal tubercles are approximately parallel. Cornicles are flanged and more than twice as long as the cauda.
Apterous: yellow green to orange, with darker patches at the base of the cornicles (Fig. 1).
Alates: yellow to brown, with dark bands on dorsal abdomen (Blackman and Eastop, 2000; Chaney and Lee, 1992).

Figure 2. Apterous foxglove aphid. Photo courtesy of UC Cooperative Extension, Monterey County, Cindy Fake 1678Hhttp://ccvipmp.ucdavis.edu/insects/foxglove.html

Adult Female: Apterous viviparous females are pale yellowish to yellowish-green or green (Fig. 1, 2), shiny; at the base of each siphunculus with a cluster of dark green or rusty-colored oily globules showing through the body. Head with prominent, parallel-sided antennal tubercles. Antennae distinctly longer than the body; segments III and IV pale with dark apices; processus terminalis about 5 times as long as the base. Abdomen without sclerotization. Siphunculi rather long and slender, cylindrical, with a very distinct flange; pale colored, but with a dark tip. Cauda pale yellowish, rather broad; a little more than one third of the length of the siphunculi (CABI, 2004).

Alate viviparous female are greenish (Fig. 3), with dark, transverse bars on the abdomen. Antennae mostly black, except for segments I and II and the basal part of III. Wing venation normal, but the anal vein (sometimes the cubital vein) much darker than the median vein. Abdominal sclerotization consisting of black transverse bars, but highly variable; from very broad bars to narrow bars of which the central portion may be missing. Legs and antennae have dark joints. Head, siphunculi and cauda are similar in appearance to apterous females (CABI, 2004).

Figure 3. Alate fox glove aphids. Photos courtesy of Robert Y, INRA Rennes (left) and Cindy Fake, UC Cooperative Extension, Monterey County (right) 1675Hhttp://www.inra.fr/Internet/Produits/HYPPZ/RAVAGEUR/3aulsol.htm, 1676Hhttp://ccvipmp.ucdavis.edu/insects/foxglove.html

Biology and Ecology

The biology of *A. solani* is complicated, as in most important aphid pest species, by the occurrence of numerous races or subspecies, including some with

particular host-plant associations (Müller, 1970, 1976). Holocyclic *A. solani* have either apterous or (more rarely) alate males, and the unusual ability to overwinter as eggs on many different host plant species, for example, on *Myosotis alpina* and on *Capsella bursa-pastoris* in Europe. Sexual forms are produced on most of these host plants. Jacob (1944) recorded adults hibernating on foxglove, and Fisken (1959) observed apterae overwintering on perennial, greenhouse, and protected horticultural crops. One generation takes about 2 weeks in favorable weather.

The epidemiology of viruses transmitted by *A. solani*, particularly potato leaf roll luteovirus and soybean dwarf virus, have been thoroughly studied in Japan in relation to its biology and flight activity (Ishitani and Niwata, 1984; Mizukoshi et al., 1991; Yamashita et al., 1991).

Pest Importance

In most potato growing areas, *A. solani* is one of the most economically important pests, causing injury either directly by their feeding punctures or indirectly by spreading viral diseases. With the use of organic insecticides, direct feeding damage has become less serious. However, more stringent permitted levels of virus infection in seed potato certification programs has increased the importance of aphids as virus vectors, since a very small percentage of infection can lead to rejection of an entire seed lot (CABI, 2004).

Symptoms/Signs

Although this aphid can infect soybean in other countries, it is more common on potato, pepper, and lettuce. Light infestations of *A. solani* can severely injure potato foliage. Its feeding causes discolored spots on tobacco, and heavily infested plants can show large necrotic areas, sometimes resulting in the senescence of the entire leaf. Feeding also causes irregular curling and distortion of young leaflets (Fig. 3). It is speculated that growth of the leaflet is hindered as a result of the feeding puncture. In potato stores, *A. solani* can attack potato sprouts.

Figure 4. Leaf distortion caused by foxglove aphid on pepper. Photo courtesy of L. Pundt, University of Connecticut. 1674Hhttp://www.hort.uconn.edu/ipm/greenhs/htms/ghsemsg200507.htm

Indirect damage is caused by honeydew production and virus transmission. Honeydew, a sticky liquid excreted by the aphid, covers the foliage, and is often colonized by black saprophytic fungi which hamper plant respiration and photosynthesis.

Known Hosts

Major hosts

Allium sativum (garlic), *Beta vulgaris var. saccharifera* (sugarbeet), *Capsicum annuum* (bell pepper), *Citrus*, *Citrus deliciosa* (mediterranean mandarin), *Citrus reticulata* (mandarin), *Citrus sinensis* (navel orange), *Cucumis sativus* (cucumber), *Fragaria ananassa* (strawberry), *Glycine max* (soybean), *Hordeum vulgare* (barley), *Lactuca sativa* (lettuce), *Lycopersicon esculentum* (tomato), *Phaseolus vulgaris* (common bean), *Polyphagous* (polyphagous), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), and *Vicia faba* (broad bean).

Minor hosts

Betula pubescens (downy birch), *Freesia*, *Lupinus* (lupines), *Nicotiana tabacum* (tobacco), *Tulipa* (tulip), and *Viola wittrockiana* (wild pansy).

Wild hosts

Achillea millefolium (yarrow), *Capsella bursa-pastoris* (shepherd's purse), *Cichorium intybus* (chicory), *Convolvulus* (morning glory), *Digitalis purpurea* (foxglove), *Plantago* (plantain), *Rumex* (dock), *Silene latifolia subsp. alba* (white campion), *Stellaria media* (common chickweed), and *Tragopogon* (goat's beard).

Known Distribution

The distribution of *A. solani* is almost worldwide. It is believed to be of European origin.

Potential Distribution Within the US

The aphid is currently present in the U.S. and is particularly common in Arizona and California. The Asian strain in the U.S. is known to occur in Hawaii and Arizona. At this time, it has not been reported as an important pest on soybean.

Survey

Aphids are efficient insect virus vectors. Thus, knowledge of aphid population dynamics is important for deciding where, when, and how to grow and protect seed potato crops. The common methods to study aphid populations are: aphids per plant counts, aphids per leaf counts, yellow water traps, and suction traps (Raman, 1985).

Aphid plant counts:

Whole plant counts, with or without beating, is a quick and efficient technique to use early in the growing season when plants are young and aphid populations are too low to be detected by other methods. The first winged adults arriving at a field usually establish aphid colonies on plants of the windward border rows. Begin the counting by randomly selecting plants along these rows. Although the number of plants to be counted depends on the size of the field, most potato workers sample 50 plants. The frequency of counting (daily to weekly) depends

on climatic conditions. Sampling should be more frequent during warm weather.

Aphid leaf counts:
Virus transmission is more closely related to the progress of aphid infestation from plant to plant, than to the total number of aphids on a particular plant. The progress of aphid infestation can be observed by the aphid per leaf counting method. In addition, natural enemies and parasitization of aphids can be observed. At least once a week, select 50 plants randomly throughout the crop. Examine three fully expanded leaves from each plant; one each on the top, middle, and lower parts. Record the number of winged and wingless aphids, as well as predators, parasites and parasitized aphids. Some winged aphids may fly away when the plant is disturbed. Nevertheless, counting aphids per leaf is more precise than aphids per plant, although it is more laborious (CABI, 2004).

Yellow water traps:
This technique has been thoroughly utilized and studied worldwide (Robert and Rouzé-Jouan, 1978). Migration of aphids into and within a potato crop is mainly due to the flight of winged adults. This flight can be studied by trapping aphids in a yellow water trap. Traps of various materials, sizes, and shapes can be used. The most frequently used traps are rectangular trays, 50 cm x 30 cm and 8 cm high, with the sides sloping outwards. Traps of the same type and size should be used in a field or region to facilitate comparison of aphid counts. If the crops are grown under rainy conditions or irrigated by sprinklers, traps need an overflow hole. A screen or piece of muslin placed over the hole allows excess water to drain without losing the aphids. One bottom corner of the tray should have an outlet tube, closed from the inside with a stopper. Two traps per field are usually sufficient. The traps should be 5 m apart from each other and situated on platforms approximately 60 cm above the ground. There should be no plants touching the platform to prevent aphids from crawling into the traps. Traps are filled with fresh, clear water 2 cm above the yellow part, with some detergent added to break surface tension and to prevent aphids from escaping. Traps should preferably be examined each morning and aphids identified with a hand lens. Suction traps have also been utilized (Taylor and Palmer, 1972; Robert, 1987; Robert et al., 1987).

Data from aphid population studies may help in forecasting the degree of virus infection. However, additional variables must be considered including: varietal susceptibility, date of planting, aphid control measures, crop management practices, and weather conditions. In general, data from aphid population studies are useful for selecting the best seed production areas, selecting the best growing season, scheduling aphicide application, scheduling dates for harvesting, and calculating action thresholds of vector populations (CABI, 2004).

A. solani is extremely polyphagous, colonizing plants in many different monocotyledonous and dicotyledonous families, with the exception of the Poaceae. Bulbs (especially tulips) often have large populations of *A. solani*, and

it is a common pest in greenhouses and on potted plants. It is common on potatoes (Blackman and Eastop, 1984). The primary hosts are hawkweed (*Hieracium* spp.) and foxglove (*Digitalis purpurea*), where this pest remains most of the year. In the summer, a small portion of the population migrates to secondary hosts. In most regions, potato is a secondary host to the foxglove aphid, where it feeds mainly on older leaves and moist areas.

Key Diagnostics

Aphids colonizing potato look similar in appearance, sharing common features such as an egg-shaped body with short cauda and antennae as long as the body. The antennal tubercles are usually prominent. *A. solani* can be recognized by its pear-shaped body, widest at the base of siphunculi with parallel sides. It has tapered siphunculi with prominent flanges and dark tips.

Apterous *A. solani* aphids range in color from light yellow-green to dark green to orange. Paler forms have a conspicuous green or rust patch at the base of each cornicle. Alates are yellow-green to brown and may have black bands on the abdomen. Both forms have prominent antennal tubercles with parallel inner faces, flanged cornicles, which are dark at the tip, and dark leg and antennae joints. The alate foxglove aphid is nearly indistinguishable from lettuce aphid in the field. See the aphid field identification guide at http://ipmworld.umn.edu/aphidalert/alert2001/Jul13/aphidkey.pdf for additional information.

Beetles/Weevils

Adoretus sinicus

Scientific Name
Adoretus sinicus Burmeister

Synonyms:
Adoretus tenuimaculatusm, Adoretus tenuimaculatus

Common Name(s)
Rose beetle, Chinese rose beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta**, Order:** Coleoptera, **Family:** Scarabaeidae

Reason for Inclusion in Manual

Pest Description

Figure 1. *A. sinicus* larva. Photo courtesy of R. Mau and J. Kessing, Department of Entomology, Hawaii. 1682Hwww.extento.hawaii.edu/kbase/crop/Type/adoretus.htm

Ovum/Egg: The small, elliptical eggs of this species are laid in the soil within 1 (2.54 cm) to ½ inch (1.27 cm) from the surface. Eggs are about 3/50 inch long by 1/25 inch wide. Appearing shining white at oviposition, the eggs gradually become dull creamy white before hatching in 7 to 16 days (Habeck, 1963).

Larvae: There are 3 larval stages of this insect that last about a week each. The larval forms of this insect are stout, 'C-shaped', white grubs with a conspicuous head and short legs (Fig. 1). The larval stage lasts for 3 to 4 weeks. Refer to Habeck (1963) for a detailed description of the larvae. Grubs do not attack live plant tissue, but preferably live in loose rich soil, leaf litter, or compost (Williams, 1931).

Pupae: Pupae are yellowish white when initially formed and gradually become brown (Fig. 2). The entire surface of the pupae is densely covered with minute hairs. The pupa is about ¼ to ½ inch long. Development is completed in 1 to 2 weeks.

Adults: The adults are sturdy, pale reddish brown beetles, and about ½ inch in length (Fig. 3). The body is covered with fine white hairs that can give the beetle a grayish appearance.

Figure 2. *A. sinicus* pupa. Photo courtesy of University of Hawaii at Hilo, College of Agriculture 1717Hhttp://www.edsung.com/roses/pests.html

Biology and Ecology

This beetle is nocturnal in habit and is attracted to lights at night. During the day they remain under leaves, loose bark, or are shallowly buried in the soil, and emerge at dusk to feed (Williams, 1931). Peak feeding and mating activity occurs about 30 minutes after sunset (Tsutsumi, et. al, 1993). Arita et al. (1993) report that this beetle preferentially feeds on leaves and plant species that are relatively high in non-structural carbohydrates. It also prefers to feed on leaves with feeding or other types of damage (Pemberton, 1959). These leaves release ethylene gas, which serve as an attractant to beetles (Arita et al., 1988). The life cycle from egg to adult is completed in 6 to 7 weeks.

Pest Importance

This polyphagous scarabaeid beetle was introduced into Hawaii sometime before 1896. It is distributed in Southeast and East Asia, including Indonesia, Taiwan, and China; it is also found on Guam. Introduction into Hawaii probably was accomplished by larvae in the soil of plants. Adults feed at night on the leaves of a great variety of plants. At least 255 plant species in 56 families have been recorded as hosts, including rose, grape, cycad, okra, beans, soybean, pigeon pea, sweet potato, eggplant, maize, cucumber, asparagus, taro, banana, and cotton. Plant damage is caused by the adult. Attacked

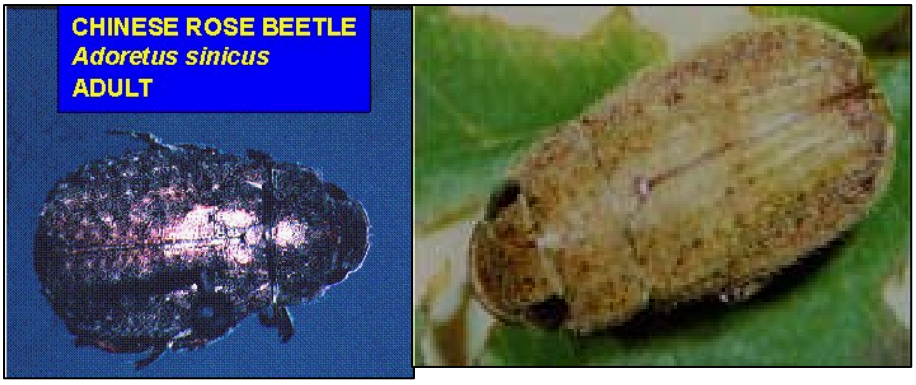


Figure 3. *A. sinicus* adult. Photos courtesy of (left) R. Mau and J. Kessing, Department of Entomology, Hawaii, (right) University of Hawaii at Hilo, College of Agriculture

leaves show numerous small holes, or may become entirely skeletonized. Larvae feed on decaying plant matter in the soil, and only rarely attack live roots. Although parasitoids and predators have been introduced into Hawaii, no satisfactory control measures have been developed for *A. sinicus*, and it remains a significant pest.

Symptoms/Signs

Adults feed on plant foliage at night, creating a lace-like or shot with holes appearance on leaves by feeding on plant tissue between leaf veins. In severe cases, most leaves are skeletonized (Fig. 4).

Larvae are commonly found in the soil of lawns, gardens, flower beds, and sometimes in cultivated fields, wherever considerable humus is present. The grubs do not attack living vegetable tissues and apparently are humus and detritus feeders.

Figure 4. Feeding damage (lace-like appearance) caused by *A. sinicus*. Photos courtesy of R. Mau and J. Kessing, Department of Entomology, Hawaii and B. Villegas 1683Hhttp://www.sactorose.org/ipm/84chineserosebeetles.htm

Known Hosts

The plant host for this species is composed of over 250 plants from a wide variety of ornamental and cultivated crops. Major crops attacked include asparagus, beans, broccoli, cabbage, cacao, Chinese broccoli, Chinese cabbage, chiso, corn, cotton, cucumber, eggplant, flowering white cabbage, ginger, grape, green bean, okra, rose, soybeans, strawberry, and sweet potato.

Major hosts

Acalypha (copperleaf), *Alocasia* (elephant ear), *Cajanus cajan* (pigeon pea), *Canna*, *Glycine max* (soybean), *Musa x paradisiaca* (plantain), *Polyphagous* (polyphagous), *Rosa* spp. (rose), and *Vitis vinifera* (grape)

Known Distribution

Originally from Japan and Taiwan, this beetle currently enjoys a widespread distribution throughout Southeast Asia and many Pacific Islands. Countries with the beetle present include China, Indonesia, Korea, Malaysia, Singapore, Thailand, Vietnam, Guam, Federated States of Micronesia, Northern Mariana Islands, and the U.S.

Potential distribution Within the US

Introduced to Hawaii before 1896, it is now a common pest on all major islands in the state. This pest is not known to occur in the continental U.S.

Survey

Surveys are conducted using a visual survey of symptoms. Look for plants with foliage demonstrating a lace-like or shot with holes appearance caused by adults of *A. sinicus* feeding on plant tissue between leaf veins. In severe cases, most leaves are skeletonized.

Key Diagnostics

Information is not available at this time.

Dectes texanus

Scientific Name
Dectes texanus LeConte

Synonyms:
Dectes texanus texanus

Common name(s)
Soybean stem borer

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Cerambycidae

Reason for Inclusion in Manual

Pest Description
The adult of the soybean stem borer (*Dectes texanus*) is a bluish grey, long horned beetle (Fig. 1). It measures about 15 mm in body length, and has prominent black and grey banded antennae, which are as long as or longer than the body length. Eggs (Fig. 1) are elongated (1.5 mm) and narrowed at both ends, shiny, yellowish, and darken to amber prior to hatching. Larvae (Fig. 2) are creamy white to yellowish, legless, cylindrical, deeply segmented, and tapered towards the rear end. The larva goes through six larval instars, and it is the larval stage that causes plant damage. The pupa resembles the adult and is about 15 mm long, yellowish when first formed, but dark brown as it matures.

Figure 1. Adult (left) and egg on leaf petiole (right) of *D. texanus*. Photos courtesy of P. Sloderbeck

Biology and Ecology

The basic biology of *D. texanus* was described in the early 1970s (Hatchett et al., 1973, 1975; Patrick, 1973). *D. texanus* overwinters as mature larvae within tunnels in the stubble of soybeans, sunflower, ragweed, cocklebur, and other weeds. Larvae pupate in early summer. Adults emerge from infested soybean stubble from mid June to early August (Daugherty and Jackson, 1969), but populations peak in July (Kaczmarek et. al., 2001). Adults live up to a month. They reach sexual maturity and are able to mate at 5 days of age. A contact sex pheromone is required for mating. Mating can occur anytime between 0900 and 1700 hours, and females can oviposit 4 to 8 days after mating (Crook et al., 2004). Eggs are laid in leaf petioles or stems in the mid-canopy (Daugherty and Jackson, 1969) (Fig. 1). When the eggs hatch, the larvae feed in the petiole for several days before tunneling down through petioles into the stem and feed on pith tissues until cold weather begins. Mature larvae girdle the stem from the inside at or below ground level and overwinter in a gallery hollowed below the girdle. Only one larva will mature in each soybean stem due to cannibalism (Sloderbeck et al., 2003).

Figure 2. Mature larva of *D. texanus*. Photo courtesy of S. D. Stewart.

In a recent study of the biology and behavior of *D. texanus* on sunflower and soybean it was found that sunflower was the preferred host plant, although females accepted soybean when it was the only available food (Michaud and Grant, 2005). It was also estimated that a single irrigated circle of sunflowers can produce up to 5 million beetles per year. Although the borers can contribute to lodging of sunflowers, their impact is probably much greater on soybean.

Soybean cultivars vary slightly in susceptibility to *D*. *texanus*, but truly resistant varieties have yet to be identified or developed (Sloderbeck et al., 2003). Later maturing varieties appear to suffer lower losses than short season cultivars. Insecticide treatments must target adults, the only exposed stage, but repeated applications may be necessary. Deep plowing, where feasible, may give some control of overwintering larvae, but is not compatible with minimum or no-till practices for dryland farming. Crop rotation is less reliable where beetle population are high. Fields with high infestation levels of *D*. *texanus* should be harvested as soon as possible to avoid girdling and lodging (Sloderbeck et al., 2003).

Pest Importance

The soybean stem borer has emerged as a major pest of soybeans only in the past 40 years (Hatchett et al., 1975; Patrick et al., 1973). Larval feeding may cause a 10% reduction in total bean weight per plant and an 8% reduction of individual bean weight (Richardson, 1975). However, lodging (breaking off) remains the primary cause of yield loss (Sloderbeck et al., 2003). Yield losses from lodging tend to be greater in early planted soybeans, non-rotated soybeans, and whenever harvesting is delayed after maturity.

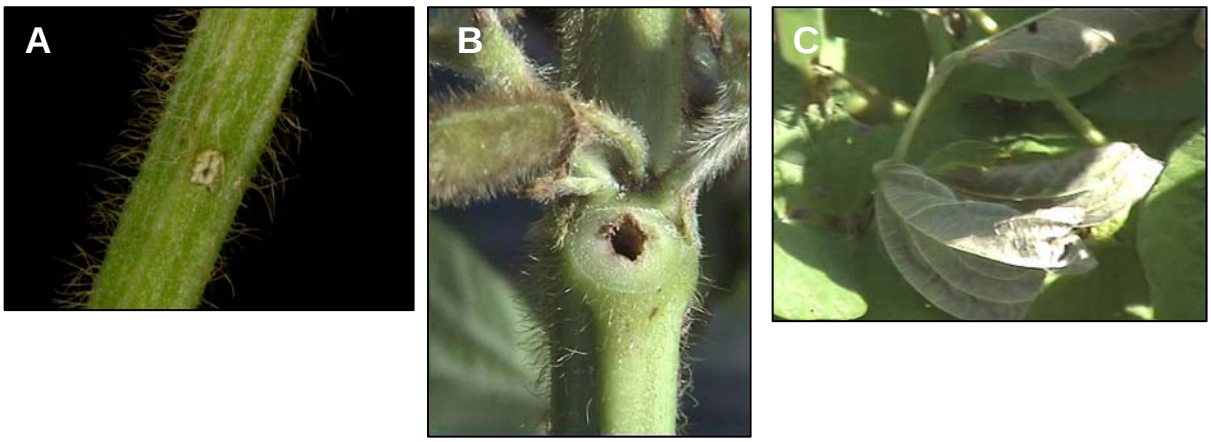


Figure 3. Oviposition scar (A), entry hole (B), and wilted leaves (C) caused by *D. texanus* Photos courtesy of S. D. Stewart.

Symptoms/Signs

Adult females chew cavities into leaf petioles or stems to deposit single eggs (Fig. 3A). Visible plant damage first becomes obvious when individual leaves begin to wilt and die (Fig. 3C) as a consequence of larvae tunneling within infested petioles. Leaves dry up and eventually drop from the plant. Wilted or dead leaves can be observed in the middle canopy of infected plants during mid summer. The reddish scar tissue in the stem that develops at the larval entrance once the wilted leaf drops off is also characteristic (Fig. 3B). If a stem is split, larva tunneling down through the stem and feeding on pith tissues can be seen (Fig. 4A). Girdled plants break easily near the soil line if subject to strong wind, heavy rain, or manual pressure (Fig. 4B). Lodged plants can be spotted in the

field later in the season and infested stubble may be visible. Girdled edges of the stem appear smooth and often show no sign of tunneling, but closer examination reveals a tunnel packed with loose fibers resembling coarse sawdust.

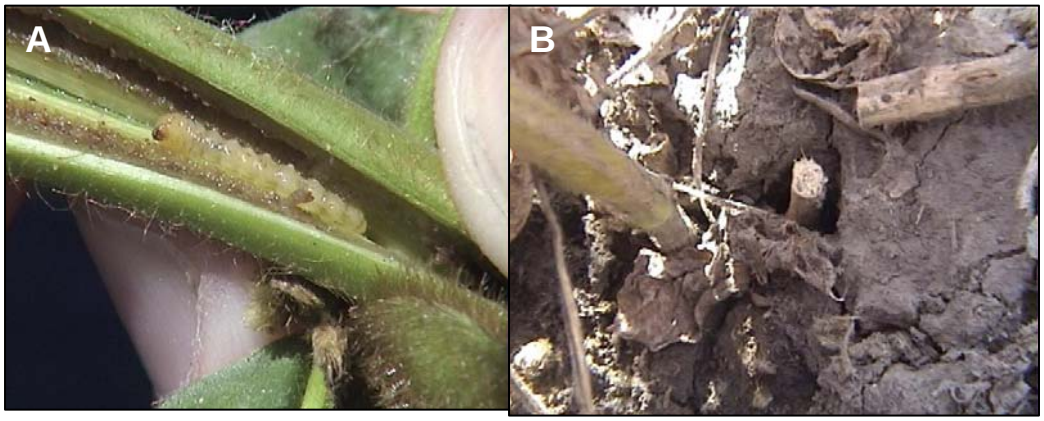


Figure 4. (A) Feeding of larva of *D. texanus* in soybean stem. (B) Girdled and lodged stem caused by *D. texanus*. Photos courtesy of P. Sloderbeck.

Known Hosts

Dectes texanus primarily attacks soybeans (*Glycine max*) and sunflower (*Helianthus annuus*). It also attacks wild sunflowers (*Helianthus* spp.), cocklebur (*Xanthium strumarium*), ragweed (*Ambrosia artemisiifolia*), and several other weeds (Hatchett, et. al., 1975).

Known Distribution/Potential Distribution Within the US

D. texanus is a native species that is widely distributed across North America east of the Rocky Mountains. It has been reported in Alabama, Arkansas, Delaware, Illinois, Kansas, Kentucky, Louisiana, Minnesota, Missouri, Nebraska, North Carolina, North Dakota, South Dakota, Tennessee, and Texas.

Survey

Infestations of soybean with *D. texanus* can be assessed by counting wilted or dead leaves in the canopy during June and July. Presence of *D. texanus* is confirmed by splitting stems and finding the tunneling larvae (Fig. 4A). Larval entry scars evident on the stem (Fig. 3B) can be used to assess incidence of infestation. Estimates of the proportion of girdled stems either in the mature crop or after harvest are useful for predicting infestation levels for the following year.

If soybean stem borer beetles are observed within the upper canopy, beetles can be sampled with a sweep net. One of the most likely places to find adults of this species is the edge rows of current soybean crop adjacent to previous year stubble. The heavier the infestation in the adjacent field, the greater the likelihood that beetles will be obvious on the new crop's foliage as they emerge from their overwintering sites in the bases of the old stems near the soil line. Fields with

sunflower the previous year are an equally important source of infestation for soybeans.

The following sampling procedure was suggested for fall sampling: In each field, 10 plants are broken at or below ground level (two plants at a time from 5 different locations). Stalks are split with a pocket knife, and examined for tunneling that starts several nodes above ground, generally where a leaf was attached. Most likely the legless, yellowish colored larvae will be near soil level during the fall season. Record the numbers of plants sampled and the percent of plants that held stem borer larvae or signs of tunnels. Also record the percent of plants that are girdled and have lodged. Larvae tend to plug the tunnel above them after they girdle a plant. Therefore, if you see lodged plants with no obvious cause, split the stems and stem bases and see if signs of tunneling and a stem borer larva can be found. Stem bases can also be split to get an idea of the infestation that had been present if the field has already been harvested. See http://www.oznet.ksu.edu/entomology/extension/Current/soybstbr.html for further details.

Key Diagnostics

Information is not available at this time.

Diabrotica speciosa

Scientific Name
Diabrotica speciosa Germar

Synonyms:
Diabrotica amabilis, Diabrotica hexaspilota, Diabrotica simoni, Diabrotica simulans, Diabrotica vigens, Galeruca speciosa

Common Name(s)
Cucurbit beetle, chrysanthemum beetle, San Antonio beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Chrysomelidae

Reason for Inclusion in Manual

Pest Description
D. speciosa was first described by Germar in 1824, as *Galeruca speciosa*. It registered another five synonyms (Araujo Marques, 1941), until the generic combination *Diabrotica speciosa* was finally proposed by Baly (1886). The subspecies *D. speciosa vigens*, is recognized from Bolivia, Peru and Ecuador, and *D. speciosa amabilis*, is found in Bolivia, Colombia, Venezuela and Panama. These two subspecies differ mainly in the coloring of the head and elytra (Araujo Marques, 1941; Bechyne and Bechyne, 1962).

Eggs: Eggs are ovoid, about 0.74 x 0.36 mm, clear white to pale yellow. They exhibit fine reticulation that under the microscope appears like a pattern of polygonal ridges that enclose a variable number of pits (12 to 30) (Krysan, 1986). Eggs are laid in the soil near the base of a host plant in clusters, lightly agglutinated by a colorless secretion. The mandibles and anal plate of the developing larvae can be seen in mature eggs.

Larvae: Defago (1991) published a detailed description of the third instar of *D. speciosa*. First instars are about 1.2 mm long, and mature third instars are about

8.5 mm long. They are subcylindrical; chalky white; head capsule dirty yellow to light brown, epicraneal and frontal sutures lighter, with long light-brown setae; mandibles reddish dark brown; antennae and palpi pale yellow. Body covered by sparse, short, dark setae; light brown irregular prothoracic plate; dark brown anal plate on the ninth segment, with a pair of small urogomphi. A pygopod is formed by the tenth segment, which serves as a locomotion and adherence organ.
Pupae: Pupae are 5.8 to 7.1 mm long; white; females with a pair of tubercles near the apex. Mature third instars build an 8 x 4 mm oval cell in the soil in which they pupate, and tenerals remain for about 3 days.

Adults: Full descriptions of *D. speciosa* are given by Baly (1886), Araujo Marques (1941), and Christensen (1943). Adults are 5.5 to 7.3 mm long; antennae 4 to 5 mm (Fig. 1). General color grass-green (USDA, 1957); antennae dark, first three basal segments lighter; head ranging from reddish brown to black; labrum, scutellum, metathorax, tibiae and tarsi black; elytra with three large oval transverse spots on each, basal spots larger and usually reddish toward the humeral callus, the rest yellow. Ventrally, head and metathorax dark brown, prothorax green, mesothorax and abdomen light brown or yellow-green. Pronotum bi-foveate, convex, smooth, shiny, ¼ wider than long. Male antennae proportionally longer than female. Males with an extra sclerite on the apex of the abdomen that makes it look blunt, compared with the rather pointed female apex.

Figure 1. Adult *D. speciosa* on soybean. Photo courtesy of CABI, 2004.

Biology and Ecology:

Eggs are laid on the soil near a host plant. Eclosion success at 27°C is approximately 92%, and it takes place after 8 days. *D. speciosa* undergoes three larval instars, which are easily differentiated by the size of the head capsule. First instars are normally scattered throughout the host's root system, but mature larvae tend to congregate in the upper 10 cm of the root under the crown. The larval stage lasts 23 to 25 days, including an inactive prepupal period of 2 to 3 days. At 25°C, the pupal stage lasts 6 days, and is followed by a period of 3 to 5 days during which the recently molted adults remain in the pupal cell, presumably for the cuticle to tan. Young beetles have a yellowish or pale brown color, which turns green with bright yellow spots in 3 days if fresh food is provided. Under laboratory conditions, mating has been observed between 4 and 6 days after emergence, and some females were observed mating again at day 35. Each female laid an average of 1164 eggs during her lifetime, starting on day 8 and extending for a maximum of 77 days. Peak oviposition was observed from days

16 to 56. The number of overlapping generations is conditioned to the climate, being continuous in tropical areas. In Buenos Aires, Argentina, observations indicate there are about three generations per year (USDA, 1957).

Pest Importance

D. speciosa is considered to be an important pest throughout southern South America (except Chile), but, being highly polyphagous, qualitative reports of its impact on different crops vary in different regions. It is considered an important pest of maize, cucurbits, and orchard crops throughout its distribution (CABI, 2004).

Adults of this chrysomelid feed on foliage, flowers and fruits of many plants. The larvae are pests of roots, especially maize. It is the most harmful species of *Diabrotica* in Argentina, especially affecting peanuts in the center of the country. It causes considerable damage to watermelon, squash and tomatoes in Brazil, and potatoes and wheat in southeast Brazil. Young squash plantings and immature tomato fruits are severely damaged in Brazil. Populations are so heavy in some years in Paraguay that vegetable crops are almost completely destroyed. Severe injury also occurs on flowers of various ornamentals such as dahlias and chrysanthemums (USDA, 1957). Economic thresholds of two insects per plant for *Phaseolus vulgaris* were determined by Pereira et al. (1997).

Symptoms/Signs

The larval damage resulting from root feeding can cause the death of the host when the host is small, but the larvae will usually only induce stunted growth in larger host plants, due to a reduction in nutrient uptake. In maize, attack on young plants produces a typical condition known as 'goose neck', in which the plant exhibits stunted growth, reduced vigor, and the first few internodes of the plant grow bent, sometimes to such an extent that the plant actually lies on the ground. In the case of peanuts and potatoes, the larvae cause external damage or short bores, similar to those of several other pests such as wireworms and other chrysomelids. The adults cause defoliation and general feeding damage to leaves, flowers and fruit. In maize, they cause a serial reduction of the number of ripening kernels from the tip of the ear to the base, due to their feeding on the tassels, which prevents pollination.

Known Hosts

D. speciosa is a highly polyphagous species as an adult, with more than 70 host species recorded (Christensen, 1943; Heineck-Leonel and Salles, 1997). The root-feeding larva is also polyphagous, but its known host range includes maize, wheat, peanut, soybeans and potato. In addition to the hosts listed, *D. speciosa* also attacks *Cucurbita maxima* subsp. *andreana* (winter squash) and *Cayaponia* species (melon leaf).

Major hosts
Arachis hypogaea (peanut), *Capsicum spp.*, *Cucurbita maxima* (giant pumpkin), *Cucurbita pepo* (ornamental gourd), *Glycine max* (soybean), *Solanum tuberosum* (potato), *Triticum* (wheat), and *Zea mays* (maize)

Minor hosts
Beta vulgaris (beetroot), *Brassicaceae* (cruciferous crops), *Citrus*, *Cucumis* (melons, cucumbers, gerkins), *Cucurbitaceae* (cucurbits), *Gossypium* (cotton), *Gossypium hirsutum* (Bourbon cotton), *Helianthus annuus* (sunflower), *Lactuca sativa* (lettuce), *Lagenaria siceraria* (bottle gourd), *Luffa aegyptiaca* (loofah), *Lycopersicon esculentum* (tomato), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Prunus persica* (peach), *Sechium edule*, *Sorghum bicolor* (sorghum), and *Triticum aestivum* (wheat)

Wild hosts
Amaranthus quitensis (pigweed (quitensis)) and *Cynara cardunculus* (cardoon)

Known Distribution
The beetle is known to occur in Central and South America. Countries reported to have the beetle present include: Costa Rica, Panama, Argentina, Bolivia, Brazil, Columbia, Ecuador, French Guiana, Paraguay, Peru, Uruguay, and Venezuela. There is a record of *D. speciosa* from Mexico, but according to Krysan (1986), it is almost certainly an error.

Potential Distribution Within the US
D. speciosa has been intercepted at ports of entry in the U.S. on several occasions (USDA, 1957), but little is known on its actual or potential distribution within the U.S.

Survey
The damage to crops from adult *D. speciosa* can be easily confused with damage by several other phytophagous insects. However, visual detection of adults is easy, as their feeding period spans from dawn until dusk. Detection of larval damage, on the other hand, is more difficult. Several larvae collecting methods and devices have been designed, but they are costly in terms of material and labor, and none have practical field use. First instars are very difficult to sample, and even large infestations can go undetected until the damage caused to the host is extensive. Larger larvae can sometimes be observed feeding on the roots of plants immediately after pulling out of the soil, but methodical sampling and counting methods have not been developed, as they have been for the North American pest species (Fisher and Bergman, 1986).

Survey and detection based on visual detection of symptoms is quite difficult and many other pests can be easily confused. Symptoms, such as dead heart in wheat, goose neck in maize, or stunted growth in most of the larval hosts of *D. speciosa*, could be attributed to several other root feeders, such as wireworms (*Conoderus* spp.; Elateridae), white grubs, (*Phytalus* spp., *Cyclocephala* spp., *Diloboderus abderus*; Melolonthidae), *Pantomorus spp.* and *Listronotus bonariensis* (Curculionidae), and several chrysomelids (*Caeporis* spp., *Colaspis spp.*, *Maecolaspis* spp., *Diphaulaca* spp. and *Cerotoma arcuata*) (Gassen, 1984, 1989).

Key Diagnostics

D. speciosa resembles somewhat the other main pestiferous Diabrotica in South America, *D. viridula*, in coloring, size, biology and host range; but *D. viridula* has dark brown areas toward the cephalic edge of the elytral spots, and distinct humeral plicae. Also, the larvae of *D. viridula* lack urogomphi on the anal plate.

Epilachna varivestis

Scientific Name
Epilachna varivestis Mulsant

Synonyms:
Epilachna corrupta, Epilachna maculiventris

Common Name(s)
Bean ladybeetle, bean ladybird, Mexican bean beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Coccinellidae

Reason for Inclusion in Manual

Pest Description
The Mexican bean beetle has a complete metamorphosis with distinct egg, larval, pupal and adult stages. Unlike most of the Coccinellidae, which are carnivorous and feed upon aphids, scales and other small insects, this species is phytophagous.

Figure 1. Egg cluster of *E. varivestis* on the underside of *Phaseolus* spp. leaf (left) and close-up of eggs (right). Photos courtesy of CABI, 2004 and J. Capinera, University of Florida.

Eggs: Dull pale yellow or orange-colored, elliptical in shape, 1.25 mm long and 0.6 mm wide, a little larger at the base (attached end) than apex, surface strongly sculptured (Fig. 1). Eggs are deposited on the lower surface of bean leaves in clusters, 6 to 75 eggs per cluster, with an average of between 40 and 50 (Chittenden and Marsh, 1920).

Larvae: Typical ladybird larva, elongate elliptical with moderately long legs, well developed head and mandibles. Yellow in color, body covered with long branched processes (scoli) bearing spines (Fig. 2). Size ranges from

approximately 1.5 mm in newly hatched larvae up to 9 mm in full-grown, fourth instar larvae (Chittenden and Marsh, 1920). A detailed morphological description was provided by Kapur (1950).

Pupae: Ovate in shape, of similar size adult, yellow with brown markings. Rather broadly rounded to subtruncate anteriorly, tapering posteriorly. Surface with sparse bristle-like setae and long hairs. Apex with two elongated processes (urogomphi), conical at base, black at extreme tips. Larval exuviae are pushed down and form a protective cover over the apical third of the body (Chittenden and Marsh, 1920). Pupation takes place in the open on an available leaf surface.

Adults: *E. varivestis* adults (Fig. 2) are 6.5 to 8 mm long. Typical ladybird beetle shape, convex dorsally, flattened ventrally, head partly hidden beneath pronotum, legs and antennae relatively short. Upper surface covered with fine, short hairs. Tarsi composed of 4 segments, second segment from base strongly lobed beneath, third segment very short and small, same width as base of claw bearing fourth segment. At low magnification, tarsus appears to be only 3-segmented, excluding tarsal claws.

Figure 2. Larvae and adult *E. varivestis*. Photos courtesy of J. Castner, University of Florida.

Body form somewhat elongate oval; elytra broadest about middle, not strongly rounded. Color variable, but generally brownish testaceous, with eight black spots on each elytron, arranged three sub-basally, three medially, and two subapically. In life, the background color is described as yellow for newly emerged adults, gradually darkening with age to a grayish brown or to a coppery color. Head and pronotum usually without spots. Darker individuals also known, elytral spots then often surrounded by pale border, or upper surface almost entirely dark brown to black. Humeral calli of elytra prominent, explanate lateral margins moderately wide anteriorly, gradually narrowing behind middle, disappearing subapically. Punctation of upper surface distinct, mixture of larger and smaller punctures on elytra; intervals mostly with weak reticulation. Tarsal claws with two long teeth.

E. varivestis and its color variations have been fully described and figured by Gordon (1975). Descriptions and illustrations were also provided by Chittenden and Marsh (1920), Howard and English (1924) and White (1941).

Biology and Ecology

The adult beetles come out of hibernation, where they have spent the winter months under collections of brush or leaves, as soon as warm weather arrives. Some may, however, delay their emergence until mid-summer. In mid-May adults tend to search out snap and lima beans, but by late June they begin ovipositing in soybeans. After feeding on the tender young bean plants for one to two weeks, the females start to lay their eggs, each depositing 500 to 600 of them in batches of 40 to 75 on the underside of the foliage. The eggs are carefully attached at the end so that they all stand vertically. They hatch in a week during warm weather but may require at least two weeks under more unfavorable conditions.

The larvae feed voraciously for two to five weeks, depending upon temperature. When first hatched, they all feed together. If the leaf is somewhat dry, the first hatched may devour the remaining unhatched eggs. As they grow older, they still retain their gregarious habits but tend to split up into small, scattered groups. When pupating, the larva fastens the tip of the abdomen to a part of the plant and starts to wiggle out of the larval skin, not entirely shedding it but pushing it back until only the tip of the abdomen remains in the skin. The pupal stage lasts for five to ten days, but may be extended in cool fall weather. The adults are strong fliers and travel long distances hunting for new bean fields. The beetles overwinter in moist, protected places, remaining dormant until spring.

Thus under favorable conditions in the southern U.S., four generations are possible each season (from April to October), whereas further north and west, only one to two generations are possible. The arrival of early autumn frosts kills many eggs, larvae and pupae in Colorado and many adults die while overwintering.

Pest Importance

E. varivestis is a major pest of beans, especially *Phaseolus* spp., in the U.S. In the early years of its spread in the eastern U.S., losses up to 100% were recorded. Figures quoted by Auclair (1960) suggested that losses of one million dollars or more annually were probably occurring in 1933. The eradication program in California cost almost one million dollars (Armitage, 1956). More recent data on crop loss due to this pest is not available.

Since the late 1960s, *E. varivestis* has been considered a major pest of soybeans in the eastern U.S., especially during more humid seasons (Elden, 1982; Kraemer et al., 1994).

Symptoms/Signs

The insect in both the larval and adult stages will feed upon the leaves, flowers and growing pods of the bean plant, but the greatest amount of injury is done to the leaves. The adult beetles are not responsible for as great a level of injury as the larvae. *E. varivestis* feeds on the underside of the bean leaves, scraping away the lower epidermal cells between the leaf veins to leave irregularly shaped

depressions or regular feeding strips. The upper epidermis is often left intact, but heavy feeding damage will completely skeletonize a leaf or leave it with a lace-like pattern (Fig. 3). Occasionally blossoms, and in many cases small pods, will be entirely destroyed or so badly eaten that they drop from the plant. Soybeans are especially vulnerable to insect defoliation during the latter period, when plants are in the podset-podfill stages. Illustrations of feeding damage were provided by Chittenden and Marsh (1920), Howard and English (1924) and White (1941).

Figure 3. Damage caused by the Mexican bean beetle. Photo courtesy of J. Caster, University of Florida.

Known Hosts

Major hosts

Citrus aurantiifolia (lime), *Glycine max* (soybean), *Phaseolus spp.* (beans), and *Vigna unguiculata* (cowpea)

Minor hosts

Lablab purpureus (hyacinth bean), *Medicago sativa* (alfalfa), and *Melilotus albus* (Bokhara clover)

Wild hosts

Desmodium (tick clovers)

Known Distribution

E. varivestis is native to Central America, having been originally described from Mexico in 1850, and probably also to the southern U.S., including Arizona, New Mexico and Colorado. Its present distribution throughout the eastern U.S. resulted from its importation to Birmingham, Alabama, probably in 1918 (Howard and English, 1924; White, 1941). From this point, it spread rapidly eastwards and northwards to the eastern seaboard and to Michigan and the New England States over the next decade, with a smaller increase in range over a second decade. The history of its spread was documented by White (1941). The present-day distribution maps (Gordon, 1975, 1985) show only a slight extension of range compared with the 1940 map of White (1941) or the CIE Map No 46 (CIE, 1954). Maes (1991) failed to find *E. varivestis* in Nicaragua and suggested that reports of its occurrence there were misidentifications.

Potential Distribution Within the U.S.

E. varivestis is already established in the majority of the U.S. *E. varivestis* is widespread in Alabama, Arizona, Colorado, Connecticut, Deleware, Georgia, Indiana, Kentucky, Maryland, New Mexico, New York, North Carolina, Ohio, Pennsylvania, South Carolina, Tennessee, and Virginia; localized in Arkansas, Florida, Idaho, Illinois, Louisiana, Maine, Massachusetts, Michigan, Mississippi, Missouri, Nebraska, New Hampshire, New Jersey, Texas, Utah, Vermont, and

Wyoming; present with few occurrences in Minnesota; and eradicated from California and South Dakota. The native habitat of *E. varivestis* has a very wet summer climate. Thus, in eastern regions with heavy precipitation, infestations are present wherever beans are grown; whereas in the west, infestations may be restricted to irrigated areas.

Survey

In fields with a history of *E. varivestis* problems, young bean plants should be monitored early in the season before immatures and adults can cause severe damage. A sweep net is considered the most economical way to sample, but a ground or shake cloth, although more cumbersome, gives more accurate counts of *E. varivestis*. Most recommendations agree that weekly sampling for insects and plant injury during the growing season is necessary to monitor populations.

Because the larvae and adults feed externally, the presence of this pest can be readily detected by observing damage to the leaves. Tapping the leaves will usually cause the beetle to fall to the ground. As the species is unlikely to occur on beans in storage conditions, its threat as a quarantine pest on beans under these conditions is remote. Accidental transportation of the adults in foliage is a more likely means for its dispersal. Howard and English (1924) thought it was possible that the appearance of the pest in Alabama in about 1919 could be due to its transportation in alfalfa hay from the southwest U.S., where the species already occurred.

Key Diagnostics

The squash beetle from North America, *Epilachna borealis*, is similar in shape and size, although slightly larger on average. It differs in having an anterior and median spot alongside the suture, common to both elytra, and by having a single apical spot (Gordon, 1976, 1985). There are many Oriental species of Epilachna, some of which are important pests, but their number and distribution of elytral spots vary from those found in *E. varivestis*.

Naupactus xanthographus

Scientific Name
Naupactus xanthographus Germar

Synonyms:
Pantomorus xanthographus

Common Name(s)
South American fruit tree weevil

Type of Pest
Weevil

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Curculionidae,

Reason for Inclusion in Manual

Pest Description
Eggs: The eggs are yellowish, ellipsoidal, and bluntly rounded at the ends; length 1.13 to 1.6 mm; width 0.4 to1 mm.

Larvae: Larvae are legless, fleshy grubs, with brown setae. The head capsule is colorless, or pigmented near the mouthparts. The mature larva is cream colored; length 14 to 18 mm, becoming 18 to 20 mm long and more globular before pupation.

Pupae: The pupa is cream-colored or slightly yellowish; length 1.1 to 2.2 cm.

Adults: Adults (Fig. 1) are flightless weevils, the males slightly smaller than the females (CABI, 2004).

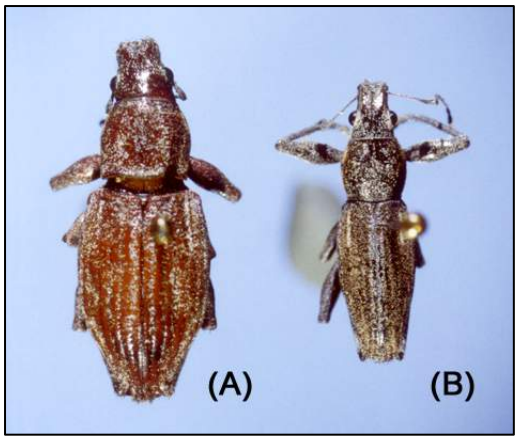


Figure 1. *N. xanthographus* adult female (A) and adult male (B). Photo courtesy of CABI, 2004.

Biology and Ecology

A generation can be completed in 19 to 20 months (Caballero, 1972). Eggs are laid at night (Gonzalez, 1980). They may be laid on the trunk of the host plant just below the branches on or under the bark, or under plastic sleeves. They are covered by a gelatinous material and laid in groups, consisting of 25 to 45 eggs (Gonzalez, 1980). In Chile, they are laid in late summer and autumn (Caballero, 1972) from January to the end of March or the beginning of April (Gonzalez, 1980). At ambient temperatures in Argentina, the incubation period ranged from 10 to 30 days during March to April and from 42 to 98 days during May to July (Loiacono and Diaz, 1992).

Eclosion of the larvae is synchronized within an egg group and occurs sometime between the end of January and the end of April (Gonzalez, 1980). The larvae are positively geotropic and enter the soil (Gonzalez, 1980), where they live at depths of 30 to 120 cm, depending on soil texture. The larval stage has five instars and lasts 11 to 14 months, or longer, but never more than 20 months (Gonzalez, 1980). The larvae feed on the rootlets of the plants or tunnel in older roots. In a greenhouse, they are known to have fed on the fine roots of the weed species provided (CABI, 2004).

Pupation occurs in the soil at depths of 30 to 80 cm (Gonzalez, 1980). Adults live for about 8 months and feed on leaves. In Chile, they remain below ground from May to September or October. They emerge from the soil over a 5 to 6 month period between spring and early autumn (Caballero, 1972), becoming most abundant in November and February. Adult emergence begins when the soil temperature at a depth of 20 cm reaches about 15 °C, and it ceases when the temperature drops below 15 °C. The peak of adult emergence occurs in late spring, a second in early summer, and a third smaller peak in late summer, mid-February to March in Chile. Females of the first peak start to lay eggs 30 to 35 days after emergence. About 24 hours after mating, females start laying eggs. They can lay up to 850 eggs and may retain viable sperm for 3.5 months. Two field-collected females laid about 490 eggs during 70 days in captivity (CABI, 2004).

Pest Importance

N. xanthographus attacks deciduous fruit trees (especially peach), vines, and other plants. It is not known to be very damaging in Uruguay. In Chile, however, it is an introduced insect and is considered one of the more important pests of grape.

Symptoms/Signs

The primary symptom is wilting of the foliage due to larval feeding. Adult feeding is noticeable only as superficial damage to leaves and fruits.

Known Hosts

Major Hosts:

Actinidia chinensis (Chinese gooseberry), *Annona cherimola* (cherimoya), *Citrus*, *Eriobotrya japonica* (loquat), *Glycine max* (soybean), *Malus domestica* (apple), *Medicago sativa* (alfalfa), *Mespilus germanica* (medlar), *Olea europaea* subsp. *europaea* (olive), *Persea americana* (avocado), *Prunus armeniaca* (apricot), *Prunus avium* (sweet cherry), *Prunus domestica* (plum), *Prunus dulcis* (almond), *Prunus persica* (peach), *Pyrus* spp. (pears), *Pyrus communis* (European pear), *Solanum tuberosum* (potato), and *Vitis vinifera* (grape)

Known Distribution

N. xanthographus is native to the southern part of South America. It has been introduced into Chile, where it is now widespread and common in the central zone (CABI, 2004).

Potential Distribution Within the U.S.

N. xanthographus has notably high pest potential for North American agriculture. It is a pest of several major crops, especially grape and tree fruits that are grown in North America. It has a primarily temperate rather than tropical distribution. Buenos Aires and Montevideo represent the general regions from which several South American weevil species (including the vegetable weevils, *Listroderes* spp., and the whitefringed weevils, *Graphognathus* spp.) probably entered southeastern North America. The weevil is immigrant in Chile, so Chilean populations should be adept at colonization. Chile should be a prime source area for immigration into western North American agricultural regions. Live adults have been intercepted at North American ports in multiple numbers. Most important, an inseminated female may retain viable sperm (Whitehead and Whittle, 1985).

Survey

Larvae can be found in the soil any month of the year. Adults can be recovered from the soil every month of the year except November in Chile, and they can be dislodged onto plastic sheets by beating the foliage. Adults cannot fly. However, they can climb the trunks of host plants, and when disturbed they drop to the ground. The legless first-instar larvae are also able to climb.

The mature larvae of *N. xanthographu*s may be mistaken for *Aegorhinus phaleratus*, which is found only in Chile, and with *Naupactus leucoloma* and *Pantomorus cervinus*.

Key Diagnostics

Adults are needed for positive identification. *N. xanthographus* and *N. dissimulator* represent two closely related species groups, which are somewhat difficult to identify to genus in existing keys to *Naupactin*i. For example, *N. xanthographus* is sexually dimorphic in several features, and in some characters (e.g., presence or absence of denticles on the ventral

margin of the hind tibia) the two sexes might trace to different genera. However, the *N. xanthographu*s and *N. dissimulator* groups are easily distinguished from all other *Naupactini* by having prominent posterior elytral tubercles formed by jointly swollen apices of intervals 3 + 9 (Whitehead and Whitten, 1985).

Popillia japonica

Scientific Name
Popillia japonica Newman

Synonyms:
Aserica japonica, Maladera japonica, Serica japonica, Autoserica japonica

Common Name(s)
Japanese beetle, velvety chafer

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Scarabaeidae

Reason for Inclusion in Manual

Pest Description
Eggs: Newly-laid eggs are about 1.5 mm long, pearly white, and oblong. Eggs absorb water from the soil, becoming spherical and nearly doubling in size within a week. The external surface of the protecting chorion is marked with small hexagonal areas. The developing embryo can be seen within eggs that are close to hatching.

Larvae: *P. japonica* larvae are typical scarabaeid grubs. The head is yellowish-brown, with strong, dark-colored mandibles; the body consists of 3 thoracic segments, each with a pair of jointed legs, and a 10-segmented abdomen. The grubs assume a C-shaped position in the soil. The cuticle is transversely wrinkled and is covered with scattered brown hairs, which are interspersed with short, blunt, brown spines. The raster, located on the ventral side of the last abdominal segment, has many scattered, brown hooked spines; medially, two conspicuous rows of 6 or 7 shorter straight spines are arranged in the form of a truncated 'V'. This V-shaped arrangement on the raster distinguishes *P. japonica* from other larval scarabaeids. The last abdominal segment also bears many yellowish hairs at the sides and the end (CABI, 2004).

Newly-hatched grubs are about 1.5 mm long and translucent white; the abdominal area becomes grayish once the larva has fed. There are three larval instars. Just prior to molting, first and second instar grubs attain average middorsal lengths of 10.5 and 18.5 mm, respectively, whereas the mature third instar averages 32 mm in length. Head capsules of first, second, and third instar grubs average 1.2 mm wide and 0.7 mm long, 1.9 mm wide and 1.2 mm long, and 3.1 mm wide and 2.1 mm long, respectively.

Prepupae: When mature, the grub stops feeding, voids the gut so that the posterior region loses its dark appearance and becomes cream-colored. It then becomes a pale and somewhat shrunken prepupa. The body straightens out except for a slight crook at the caudal end. Eventually, the developing appendages are everted from their sacs and lie outside the newly developed pupal cuticula, beneath the old larval cuticula. The transformation to prepupa and pupa occurs in the earthen cell formed by the mature larva (CABI, 2004).

Pupae: The young pupa forms within the old larval and prepupal exuviae, which change in appearance to a fine, light tan mesh-like tissue. This shroud-like covering splits along the middorsal line as the pupa develops. The pupa, which averages 14 mm long and 7 mm wide, resembles somewhat the adult beetle, except that the wings and other appendages are closely folded to the body. It is a pale cream color at first, gradually becoming tan and finally taking on the metallic green of the adult.

Figure 1. Adult Japanese Beetle. Photo courtesy of Doug Stone, Mississippi State University, 1684Hwww.invasive.org

Adults: The adult is an attractive, broadly oval beetle, 8 to 11 mm long, and about 5 to 7 mm wide. Females usually are slightly larger than males. The head and body are dark, metallic green, with darker coppery-green legs (Fig. 1). The coppery-brown elytra, which do not quite reach the tip of the abdomen, expose a row of 5 lateral patches of white hairs on each side of the abdomen and a pair of these patches on the dorsal surface of the last abdominal segment. These white patches on the green abdomen distinguish *P. japonica* from all other beetles that resemble it (CABI, 2004).

Biology and Ecology

Larvae (usually 3rd instar) overwinter in an earthen cell about 15 to 30 cm deep in the soil. In early spring, when the soil temperatures warm to about 10°C, the grubs move closer to the surface and resume feeding on plant roots at 2.5 to 5 cm depth. Pupation usually occurs after 4 to 6 weeks of feeding, and the adults emerge in late May to early July, depending on latitude (CABI, 2004).

Mating and egg-laying begin soon after emergence. Virgin females produce a volatile sex pheromone. Early in the seasonal flight period, aggregations containing dozens of males may form on the ground around a single, emerging female. Females also re-mate on food plants between each bout of oviposition. The beetles usually feed in groups, usually starting near the top of a plant and working downward. The adults are attracted to feeding-induced plant volatiles, resulting in aggregation on damaged plants. Females may leave host plants in the afternoon and fly to suitable sites for oviposition. Areas with moist, loamy soil covered with turf or pasture grasses are preferred, especially when such sites are near favored food plants. Eggs are laid singly, in small clutches (1 to 4 eggs) in the upper 7.5 cm of soil. The cycle of feeding and oviposition is repeated every few days. The average life of a female is 30 to 45 days, during which she may lay 40 to 60 eggs (CABI, 2004).

Eggs hatch in about 2 weeks, and the young grubs begin feeding on fine roots and organic matter. They molt and become second instars after 2 to 3 weeks, and third instars after 3 to 4 weeks more. Feeding continues until late fall, when the grubs move deeper into the soil in response to declining soil temperatures. At the latitude of Virginia and Maryland, the population consists of mainly adults and eggs in July, first and second-instar grubs by mid-August, second and third instars by early September, third instars from late September to late April, and prepupae and pupae in May and early June. This sequence is 2 to 3 weeks later in more northern parts of the beetles' range, and somewhat earlier in the south. Normally there is one generation per year, but at the northern edge of its range, a few individuals may need 2 years to complete the life cycle (CABI, 2004).

Temperature and soil moisture are probably the main factors limiting potential spread of the beetle into new areas. According to Fleming (1972), *P. japonica* is adapted to regions where the mean soil temperature is between 17.5 and 27.5°C during the summer and above -9.4°C in winter. In addition, precipitation must be adequate and rather uniformly distributed throughout the year, averaging at least 25 cm during the summer.

Pest Importance

P. japonica is probably the single most destructive insect pest of golf courses, lawns, and herbaceous and woody landscape plants in the eastern U.S. Hundreds of millions of U.S. dollars are expended each year in controlling the grubs and adults, and in renovating or replacing damaged turf or ornamental plants. Damage to tree fruits, small fruits, maize and soybeans is also significant. Many millions of U.S. dollars have also been spent in limiting the beetles' spread in North America. It is less of a pest in Japan.

In the U.S., adult *P. japonica* have been observed feeding on at least 295 species of plants in 79 plant families. These include small fruits, tree fruits, vegetable and garden crops, field crops, woody and herbaceous ornamentals, shade trees, various weeds, and many non-economic species. Economic

damage has been recorded on more than 100 species. The beetles are particularly attracted to certain species of Aceraceae, Anacardiaceae, Betulaceae, Clethraceae, Ericaceae, Fagaceae, Gramineae, Hippocastanaceae, Juglandaceae, Lauraceae, Leguminosae, Liliaceae, Lythraceae, Lythraceae, Malvaceae, Onagraceae, Platanaceae, Polygonaceae, Rosaceae, Salicaceae, Tiliaceae, Ulmaceae, and Vitaceae. The grubs feed on roots of a wide range of vegetable crops, ornamental plants and tender grasses. In Japan, the host range appears to be smaller than in North America.

Symptoms/Signs

Skeletonized foliage is the most common symptom of feeding by the adult (Fig. 2). The beetles generally feed from the upper surface of leaves, chewing out the tissue between the veins and leaving a lacelike skeleton. Severely damaged leaves soon turn brown and drop. The adults are gregarious, usually beginning to feed on foliage at the top of a plant and working downward. On some plants with thin leaves and fine venation, and on petals of flowers (Fig. 2), the beetles consume irregularly shaped sections in the same manner as a caterpillar. Plants with thick, tough leaves are usually not attacked, but when such leaves are eaten, the feeding often is restricted to the palisade mesophyll and does not penetrate to the lower leaf surface.

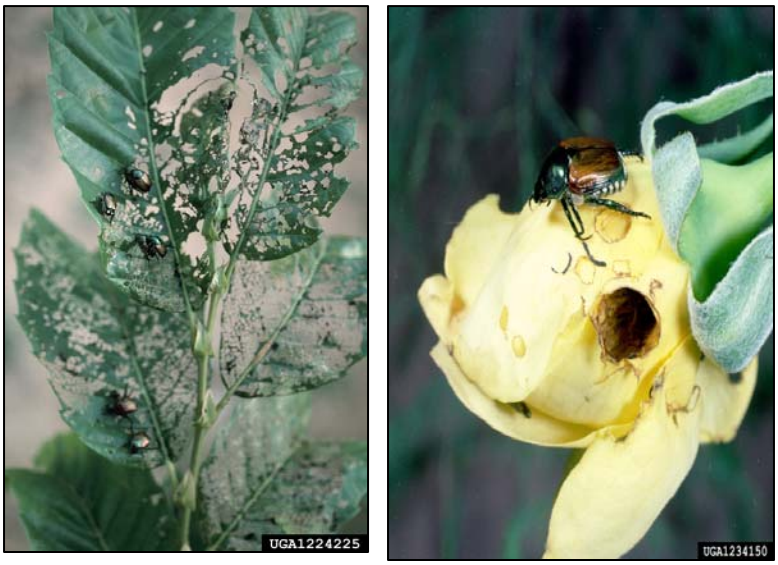


Figure 2. Japanese beetle damage. Photos courtesy of Jerry A. Payne, Agricultural Research Service and Clemson University. 1685Hwww.invasive.org

Maize is the field crop most seriously damaged in North America. The beetles feed on the maturing silk, preventing pollination; this results in malformed kernels and reduced yield. They also defoliate soybeans, asparagus, all varieties of grapes, and various fruit-bearing trees, especially apple, cherry and plum. Beetles often aggregate and feed in large numbers on fruit of early-ripening varieties of apple, peach, nectarine, plum and quince, rendering them unmarketable.

The larvae are most abundant in well-kept lawns and golf courses, and in pastures. As the grub burrows through the soil just below the surface, it cuts off and consumes the grass roots. Early symptoms include thinning, yellowing, and wilting, culminating in large patches of dead, brown grass that appear in late

summer or early autumn, and sometimes in the following spring. When grubs are numerous, the root system is completely severed, and the sod can be lifted or rolled back like a carpet. Damage is compounded by deficiency of rainfall or other stress. Secondary damage from skunks, raccoons, birds, moles or other predators often causes more disruption to the sward than the grubs themselves. Feeding by grubs on roots of maize, beans, tomatoes, strawberries, nursery seedlings or other crops reduces their vitality and yield and sometimes kills the plants (CABI, 2004).

Known Hosts

Major hosts

Acer (maples), *Asparagus officinalis* (asparagus), *Glycine max* (soybean), *Malus* (ornamental species apple), *Prunus* (stone fruit), *Rheum hybridum* (rhubarb), *Rosa* (roses), *Rubus* (blackberry, raspberry), *Tilia* spp.(limes), *Ulmus* (elms), *Vitis* (grapes), and *Zea mays* (maize).

Minor hosts

Aesculus (buckeye), *Althaea* (hollyhocks), *Betula* (birches), *Castanea* (chestnuts), *Hibiscus* (rosemallows), *Juglans nigra* (black walnut), *Platanus* (planes), *Populu*s (poplars), *Salix* spp. (willow), *Sassafras albidum* (common sassafras), *Sorbus americana* (American mountainash), and turfgrasses (CABI, 2004).

Known Distribution

P. japonica originates from northeastern Asia where it is native in northern China, Japan and in the far east of Russia. The pest also occurs in the U.S. In Japan, the beetle is most abundant in northern Honshu and in all of Hokkaido where grasslands occur, but it rarely reaches the high population densities that occur in the eastern U.S. In Russia, *P. japonica* presently is restricted to the South Kirile region of Sakhalin, on the island of Kunashir. It is absent in the European Plant Protection Organization (EPPO) region, except for the island of Terceira, Azores (Portugal), where the pest has spread from a U.S. airbase. The pest has a localized distribution in Canada (Nova Scotia, Ontario, and Quebec) (CABI, 2004).

Potential Distribution Within the U.S.

In the U.S., *P. japonic*a is now established in all eastern states except for Florida. *P. japonica* is present in Alabama, Connecticut, Delaware, Georgia, Idaho, Illinois, Indiana, Iowa, Kansas, Kentucky, Maine, Maryland, Massachusetts, Michigan, Minnesota, Missouri, Nebraska, New Hampshire, New Jersey, New York, North Carolina, Ohio, Pennsylvania, Rhode Island, South Carolina, Tennessee, Vermont, Virginia, West Virginia, and Wisconsin. The pest is believed to be eradicated in California, Nevada, Oregon, and Washington.

Allsopp (1996) used a computer-generated modified Match Index to analyze climatic suitability and predict the potential worldwide distribution of *P. japonica.*

According to the model, in North America, the beetle has the potential to spread west to the middle of Nebraska, Kansas, Oklahoma and Texas; south to the middle of South Carolina and Georgia and most of Alabama and Mississippi; it already has reached some of these limits. The southern parts of the Canadian Maritimes and eastern British Columbia and parts of Washington and Oregon are also suitable.

Survey

Adult *P. japonica* are easily detected by inspecting vulnerable plants for aggregations of beetles on foliage, flowers, or fruits, or for skeletonized leaves, during the beetles' flight period in early- to mid-summer. Adults are most active on warm sunny days. Traps containing food-type lures and/or sex attractants have been widely used in the U.S. for monitoring and survey purposes, and to detect infestations. Active grubs can be detected by cutting sections of sod with a spade or golf-type cup cutter in late summer or autumn, or in early spring, and examining the soil and roots to a depth of about 8 cm.

Key Diagnostics

The adults are a brilliant metallic green, generally oval in outline, 3/8 inch (8 to 11 mm) long and 1/4 inch (5 to 7 mm) wide. The wing covers are a coppery color and the abdomen has a row of five tufts of white hairs on each side that are diagnostic.

Flies

Melanagromyza dolichostigma

Scientific Name
Melanagromyza dolichostigma De Meijere

Synonyms:
Agromyza dolichostigma, Melanagromyza decora

Common Name(s)
Soybean root miner

Type of Pest
Fly

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Agromyzidae

Reason for Inclusion in Manual

Pest Description
Generally speaking, agromyzid flies are small and black or yellowish (Fig. 1). Larvae are leaf miners, while adults can be found in a diversity of settings. Most plant damage is accomplished by fly larvae. Larval mine characters often afford the easiest recognition of agromyzid flies, rather than the individuals themselves. These are usually serpentine mines: narrow and winding, which increase in width with larval growth (Triplehorn and Johnson, 2005). Other species of agromyzids may instead form blotch or intermediate mines (Baker and Bambara, 1997).

Figure 1. Agromyzid fly adult. Photo courtesy of AVRDC Crop Protection Guides

In 1922, De Meijere described *M. dolichostigma*

from a series of specimens and *M. decora* from a single female, both from material bred from *Glycine* (as Soja) and *Phaseolus*. *M. decora* was later synonymized with *M. dolichostigma* (Spencer, 1961).

Eggs: Eggs are yellowish-white and cylindrical. The surface is marked with distinct longitudinal grooves.

Larvae: Larvae are long and slender with teeth of differing length on each mouth-hook; anterior spiracles unusually long, with up to 12 minute pores; posterior spiracles shorter, but still elongate with up to 15 pores; color off-white.

Pupae: Pupae are oval, 2.5 mm long, yellowish brown in color and remain in stems.

Adults: *M. dolichostigma* is a small blackish species with a variable metallic coloring, usually of a copper/purple hue. Frons narrow; jowls narrow; eyes bare; third antennal segment small, round and with a noticeably long and pubescent arista. Mesonotum with two strong dorso-central bristles and between them, numerous rows of fine acrostical hairs (up to 8 rows). Mid-tibiae of the legs with two strong lateral bristles. Wing length ranges from 1.8 mm in male to 2.2 mm in female; costa extending strongly to M<(sub)1+2> (last long vein to reach apex of wing). Last long vein, M<(sub)3+4>, with a cross-vein dividing it into proximal one-third to distal two-thirds. Head, frons, matte black; orbits and ocellar triangle, only slightly shining; thorax and abdomen with a metallic sheen of green to purple; squamae pale with a yellow-brown margin. Details of genitalia are given in Spencer (1973).

Biology and Ecology

According to van der Goot (1930), eggs are laid exclusively on the underside of leaves. Frequently, the eggs are not inserted into the leaf tissue but are merely deposited on the surface. Feeding punctures, however, are made on the upperside of the leaves. Three or four eggs may be laid together on one leaf. Leaves are usually selected which have not fully unfolded and this, together with the hairs on the underside, prevents the eggs from falling to the ground before the larvae hatch. Hatching takes place within 2 to 3 days of oviposition.

Upon hatching, the larva immediately eats its way into the leaf tissue and then into the nearest vein and via the petiole to the stem. Here it feeds initially on the outer layers. After feeding down the stem for 2 to 3 cm, the larva turns and feeds upwards eating deeper in the pith of the stem. A substantial cavity is eaten out, enclosed only by the outer layers of the stem tissue and the epidermis.

Frequently, two or more larvae can be found feeding together in *Phaseolus vulgaris*. This is particularly common in smaller plants, where the entire stem tissue is eaten and the shoot dies as a result. In more robust plants, the shoot is able to continue its growth, and a gall-like swelling develops at the point of larval

feeding. This regularly occurs in *Vigna umbellata* var. *trinervis*. The duration of larval feeding has not been accurately observed but appears to be 8 to 10 days.

Pupation takes place in the hollowed-out stem. The puparia are always in the upper-most part of the stem, either in the shoot that has been killed or in the swelling immediately beneath the shoot. On one occasion, puparia were found in the swollen petiole of one of the upper leaves.

The duration of the entire life cycle was studied in the laboratory and has ranged 17 to 21 days, averaging 18 days. Females lived in the laboratory for 11 to 44 days, averaging 22 days.

Pest Importance

Although individual larvae damage can be high, *M. dolichostigma* is regarded as not very dangerous because populations are normally smaller than those of other agromyzids on tropical legumes (Spencer, 1973). However, in mountain areas of Java and Taiwan the damage can be more significant (Lee, 1976; van der Goot, 1930). Talekar (1990) reported that the damage of *M. dolichostigma* is greater after the flowering period of the host plant. After flowering, the plants can hardly compensate for wilted shoots.

For various reasons, the damage caused by *M. dolichostigma* is generally not serious. Populations of *M. dolichostigma* are generally low. Frequently, the first attack takes place shortly before flowering and when the main growth has been completed. Despite damage, and even death of a short length of the shoot, the plant frequently survives and is able to resume normal growth. However, when 3- or 4-week-old plants are attacked, stunting of growth occurs, and this is rarely compensated by production of new shoots. The production of pods and the yield is then considerably reduced, as was seen with a crop of soybean in Lembang, Java (Indonesia), in 1921. In this case about 50% of plants were attacked. Normally, however, the infestation rate is approximately 1 to 2%. On *Vigna umbellata,* the frequent death of the top shoots inhibits growth and leads to a reduced number of pods. Infestation occurs regularly at a low level, but on occasions it is severe. On this evidence *M. dolichostigma* cannot be considered a serious pest. Nevertheless, when populations rise due to favorable circumstances economic loss is clearly caused (Spencer, 1973).

Since 1973, the area of soybean cultivation has considerably increased. For instance, in Japan, government efforts are being made to increase the production of soybean at the expense of rice growing. Also, in Taiwan, more soybeans are being grown than 29 years ago (Chang, 1971). Thus, in Japan and Taiwan, the presence of *M. dolichostigma* can pose a threat to the increased acreage of soybean.

Symptoms/Signs
Larvae feed internally. In smaller plants, the entire stem tissue may be eaten and the shoot may die as a result. In more robust plants, the shoot is able to continue its growth and a gall-like swelling develops at the point of larval feeding. The whole plant may wilt and stunt (dwarf). The production of pods and the yield can be considerably reduced.

Known Hosts
Major hosts
Cajanus (pigeon pea), *Fabaceae* (leguminous plants), *Glycine max* (soybean), *Pachyrrhizus*, *Phaseolus* (beans), *Psophocarpus* (winged bean), and *Vigna* (cowpea). The main host appears to be soybean.

Minor hosts
Arachis (peanut), *Calopogonium* (calopo), *Canavalia* (jack bean), *Centrosema pubescens* (centro), *Crotalaria* (rattle pod), *Indigofera* (indigo), *Pueraria* (kudzu), and *Rhynchosia*.

Known Distribution
*M. dolichostigma*a seems to be restricted to Southeast Asia, where it is thought to be indigenous. The fly has been reported in China, Taiwan, Java (Indonesia), Thailand, and Japan. These are the only recorded areas for *M. dolichostigma* despite the fact that it infests such a widely grown host as soybean. This would suggest that the fly is naturally restricted to its indigenous countries. Elsewhere, soybean and the other legumes reported as hosts for *M. dolichostigma* are attacked by other pests, including other *Melanagromyza* species.

Potential Distribution Within the US
Information is not available at this time.

Survey
Inspect young leaves, in particular, for signs of egg laying by *M. dolichostigma* or for larval activity in the leaf, petiole or stem. Later symptoms to look for are swollen stem and dead leaves associated with dead shoots.

Key Diagnostics
M. dolichostigma has similarity to several other related species, thus reliable identification is not easy.

The three species *M. bonavistae* in East Africa, *M. dolichostigma* in Java and Formosa, and *M. koizumii* in Japan represent a closely related complex, as seen from the unusually long larval anterior spiracles and the characteristic form of the posterior spiracles. Apart from the differences in genitalia, there are also significant differences in biology; *M. bonavistae* feeds in pods, *M. dolichostigma* and *M. koizumii* feed in stems (Spencer, 1973).

The larvae lack the typical central horn of posterior spiracles. Larval mandibles have one mouth hook each, the left one distinctly larger than the right one. Locomotion welts clearly consist of two types of denticles (Sasakawa, 1961). Two striking characters are the extremely elongated anterior and posterior spiracles. However, these features are shared with the related but only poorly understood species *M. koizumii* from Japan. Separation of the two species is possible only by the mode of female oviposition.

The male genitalia of *M. dolichostigma* are similar to *M. cordiophoet*a, *M. sojae,* and *M*. *cunctans*; however, other morphological characters and some host preferences are different. Whilst all *Melanagromyza* share the metallic coloring to some degree, *M. dolichostigma* is quite distinct in having the strong purple tinge to its color.

Melanagromyza sojae

Scientific Name
Melanagromyza sojae Zehntner

Synonyms:
Agromyza producta, Agromyza sojae, Melanagromyza producta, *Melanagromyza prolifica*

Common Name(s)
Asparagus miner, bean stem miner, bean fly, stem fly, soybean fly, soybean stem borer, soybean stem miner, soybean stem fly

Type of Pest
Fly

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Agromyzidae

Reason for Inclusion in Manual

Pest Description
The egg is whitish, partly transparent and measures 0.34 ± 0.02 mm in length and 0.15 ± 0.01 mm in width (Lee, 1976; Wang, 1979).

The young larva is nearly colorless (Fig. 1). The peculiar shape, size and nature of sclerotization of posterior spiracular bulbs can be used in identification. The anterior spiracles are short and knoblike, with eight minute pores. Posterior spiracles are well separated and normally consist of six raised pores around a central truncated horn. The three larval instars of the soybean stemfly, *M. sojae*, are completed in 9 to 11 days.

Figure 1. Larvae (left) and pupa (right) of *M. sojae*. Photo courtesy of Ooi. P. 1686Hwww.ecoport.com

The pupa is cylindrical, golden yellow, and measures 2.75 mm long and 1.00 mm wide (Singh, 1982) (Fig. 1). Pupation of the soybean stem fly occurs in the center of the bean stem, often at the level of the unifoliate leaves of younger plants.

Freshly emerged adult flies have moist crumpled wings and very faint pigmentation on the abdomen and legs. Progressive darkening and hardening of the body wall and legs occurs for about 30 minutes, during which the wings also become smooth and dry. Soon the fly develops its metallic black color with a metallic shiny abdomen. Antennae, legs, and bristles on head and thorax are all black. The wings are transparent. Females are larger and have a tube-shaped abdomen. In females, body length is 1.88 mm, width at thorax 0.70 mm, wingspan 4.45 mm. In males, body length is 1.60 mm, width at thorax 0.50 mm, and wingspan 3.90 mm. Spencer (1973) gives details of other morphological characters.

Biology and Ecology

Eggs are always laid on the underside of the young leaves; on a unifoliate leaf if the plant has only two leaves, or on fully opened trifoliate leaves, at the basal part of the leaf lamina near the petiole. Numerous feeding punctures are made on the upper side of the leaves. One leaflet usually receives 1 or 2 eggs; however, that number may reach 5 or 6 depending upon adult population density. Egg hatch commences in 2 days, peaks in 3 days and can last up to 7 days after oviposition (Wang, 1979).

Immediately after emergence, the larva burrows through the mesophyll tissue into the closest vein, disappearing downwards in the leaf, eventually tunneling through the petiole and ending up in the stem. In the stem, the larva burrows into the pith reaching the root-shoot junction. It burrows further into the thickened tap root, turns around, and moves upward into the pith, thus widening the original tunnel. It gnaws through xylem and phloem tissues to the epidermis, making a hole to the outside, closes it with debris, and pupates in the stem (van der Goot, 1930).

The larva is nearly colorless and attracts very little attention when the stem is cut open for observation. There are three larval instars. Singh (1982) reports the duration of three instars at 32 ± 2°C and 70% RH as follows: first instar 22 hours, second instar 43 hours and third instar 98 hours. The total duration of larval stage was 7 days. Natural mortality of larvae is very high. Despite the large number of eggs, a maximum of only two larvae were found in van der Goot's (1930) study in Indonesia. Wang (1979) reports 62, 24, and 20% mortality of larvae in 1st, 2nd, and 3rd instars, respectively.

The pupa is always located in the pith tunnel, often at the level of unifoliate leaves of younger plants and usually near the fly escape hole, seen as a dark depression. The duration of the pupal period in the laboratory at 30 ± 2°C and 70% RH was 190 hours (Singh, 1982). At an average temperature of 27°C, the

pupal stage lasts 6 to 9 days in June in northern Taiwan (Lee, 1976). In Indonesia, van der Goot (1930) reported a pupal period of 9 to 10 days.

The majority of adults emerge during the morning and early hours of the day. The total development time from egg to adult is 16 to 26 days, with an average of 21 days, in lowland Indonesia. Soon, the fly develops its metallic black color and seeks soybean and other host plants. *M. sojae* adults are weak fliers and their activity is strongly influenced by the weather. They feed on plant juices from oviposition and feeding holes made in the leaves by females, dew drops, and other similar moist materials. Copulation occurs 3 to 5 days after adult emergence. The insect copulates only in the morning from 0700 to 1000 hours. Oviposition begins soon after copulation and lasts for 19 days (Wang, 1979). Eggs are laid on the leaves. In Taiwan, females laid 171 ± 115 eggs throughout their lives. The females each laid 1 to 34 eggs per day, and 50% of eggs were laid within the first 9 days (Wang, 1979).

Van der Goot (1930) found the lifespan of adults in the laboratory to be 15 to 36 days with an average of 23 days for females and 10 to 46 days with an average of 26 days for males. This lifespan, according to the same study, was longer than it is under field conditions. In Taiwan, Wang (1979) reports the lifespan as 6 to 19 days for adult flies. In India, Singh (1982) reports the average lifespan as slightly more than 4 days at 30 ± 2°C and 70% RH.

Pest Importance

M. sojae is a pest mainly of soybean and to some extent mung bean and black gram. In soybean, infestation occurs in the unifoliate or early trifoliate leaf stage. By this stage, the seedlings are well established, and the insect infestation rarely results in plant mortality. Yield loss varies from location to location and according to the plant growth stage when infestation occurs. Yield reduction occurs only when the plant is damaged at the seedling stage. The later the damage, the lower the yield loss will be. In Taiwan, yield loss among 163 soybean varieties was 31% (AVRDC, 1981). In Shandong Province of China, there are reports of *M. sojae* causing plant mortality and yield loss in soybean (Anonymous, 1978). In India, Bhattarcharjee (1980) studied the relationship between *M. sojae* infestations, plant height and yield loss in soybean. According to his calculations, this insect has the potential to cause up to 80% yield loss. This pest probably causes significant yield loss in soybean in Indonesia. However, in most cases, if the crop is not protected,

Figure 2. Larval stem borings caused by *M. sojae*. Photo courtesy of CABI, 2004.

Ophiomyia phaseoli causes severe damage before *M. sojae* infestation begins. Hence, no independent information is available on the extent of plant damage or yield loss by *M. sojae* in that country.

Symptoms/Signs

M. sojae overwhelmingly prefers soybean. There are no clear external symptoms of infestation except for some minute oviposition/feeding punctures at the base of the leaf lamina. When the stem is cut open, feeding tunnels containing larvae and pupae are visible (Fig. 2). In slightly older plants, two separate tunnels are often found. The one in the lower half is older and has developed a dark brown color. This tunnel originates in the stem, roughly at the junction of the unifoliate leaves, and extends downwards up to the soil surface. This type of tunnel indicates that the infestation occurred earlier, from the eggs laid in the unifoliate leaf. The second tunnel starts just under the top of the plant and extends downwards up to the first tunnel. Presuming that the part of the plant at the unifoliate leaf escaped infestation, this tunnel can extend up to the soil surface. This feeding results from the later infestation of trifoliate leaves. If the plant is damaged very early, it is possible that the later infesting larvae will not have enough pith tissue to feed on. Under such circumstances, the larva moves upwards hollowing out the shoot, which may lead to withering of the top.

Known Hosts

Major hosts

Glycine max (soybean)

Minor hosts

Cajanus cajan (pigeon pea), *Crotalaria juncea* (sunn hemp), *Medicago sativa* (alfalfa), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Vigna angularis* (adzuki bean), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), *Vigna sinensis ssp. sesquipedalis* (asparagus bean), and *Vigna unguiculata* (cowpea)

Wild hosts

Astragalus sinicus

Known Distribution

M. sojae is also widespread in China, India, Indonesia, Japan, Korea, and Vietnam. It is present in Israel, Laos, Malaysia, Philippines, Saudi Arabia, Thailand, Egypt, South Africa, Australia, and the Solomon Islands.

Potential Distribution Within the US

Information is not available at this time.

Survey

M. sojae is metallic black in color, 1.9 mm in length, and shaped like a minute fly. Adults are active only during the day, especially in the early morning hours. In

young leaves, *M. sojae* causes tiny holes at the base of the leaf lamina. On cutting open, stems are seen to contain dark red to brownish feeding damage in the pith and larvae or pupae in the feeding tunnels.

Key Diagnostics

Two other agromyzids, *Ophiomyia phaseoli* and *O. centrosematis*, can attack soybean at the same time as *M. sojae*. These insects can be easily distinguished only in larval and pupal stages by locating their feeding and pupation sites within the host plant. Whereas *Ophiomyia* larvae are cortex feeders, pupating in the cortex just beneath the stem epidermis, *M. sojae* larvae are pith feeders and pupate in the pith. The three species can also be distinguished by the morphology of their posterior spiracles in both larval and pupal stages (Talekar, 1990). In *O. phaseoli,* the posterior spiracles closely adjoin on conical projections usually with about 10 minute bulbs; in *O. centrosematis*, the distal ends of the posterior spiracles are divided into three conical structures; and in *M. sojae*, they are well separated and normally consist of six raised pores around a central truncated horn.

Ophiomyia centrosematis

Scientific names
Ophiomyia centrosematis de Meijere

Synonyms:
Melanagromyza centrosematis

Common Name(s)
Bean root miner, stemfly

Type of Pest
Fly

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Agromyzidae

Reason for Inclusion in Manual

Pest Description
The eggs of *O. centrosematis* are laid underneath the epidermis in the hypocotyls of the host plant. The almost transparent eggs are 0.413 ± 0.023 mm long and 0.163 ± 0.025 mm wide (CPC Report, 1997).

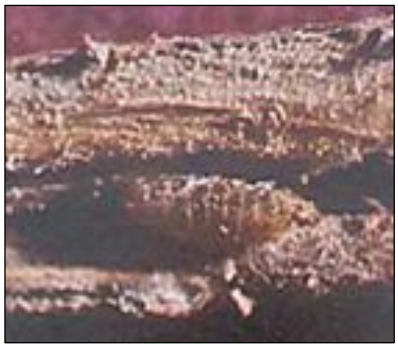


Figure 1. Agromyzid fly pupa inside stem. Photo courtey of AVRDC Crop Protection Guides.

There are three larval instars. The first instar is almost transparent; while the second and third instars are milky white. Larvae become opaque before pupation. The length of the cephalopharyngeal apparatus was 0.22 ± 0.03 mm in the first instar, 0.44 ± 0.02 mm in the second, and 0.64 ± 0.02 mm in the third (Talekar and Lee, 1988). There is, therefore, linear increase in the length of the cephalopharyngeal apparatus from the first through the third instar. The anterior spiracles are much longer than the posterior spiracles. The distal end of the posterior spiracle is divided into three

conical structures with one opening on each (Talekar, 1990). This feature is retained in pupae (CPC Report, 1997).

Initially, pupae (Fig. 1) are light yellow, becoming golden yellow and dark yellow just before adult emergence. Pupae, on average, are 2.30 ± 0.10 mm long, 0.89 ± 0.07 mm wide, and weigh 0.708 + 0.021 mg (Talekar and Lee, 1988).

Adults are small, shiny and black. Spencer (1973) describes details of other morphological characters.

Biology and Ecology

The life cycle of beanflies is completed very rapidly, often in less than 2 weeks. Generations are continual in tropical areas. Pupation occurs inside the stem and adult egg-laying activity occurs mainly in the leaves near the petiole (AVRDC Crop Protection Guides, 2001). Temperature strongly influences all growth stages; warmer temperatures correspond with shorter egg, larval, pupal and adult stages. Three to four generations of *O. centrosematis* are typically observed in Taiwan, where the average temperature is 25°C (Talekar and Lee, 1988). However, in Taiwan, yield losses as a result of agromyzid fly damage typically only occur within 4 weeks of soybean germination, correlating with the first two generations of the fly only.

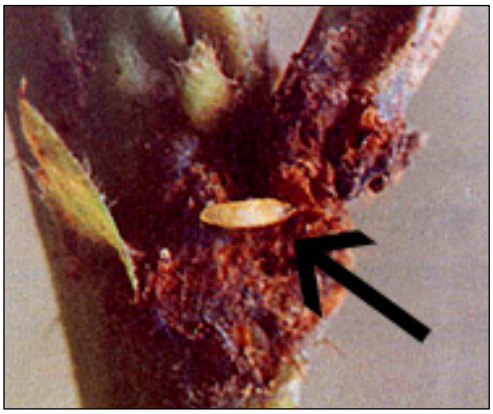


Figure 2. Agromyzid fly larva. Photo courtesy of AVRDC Crop Protection Guides.

Females lay eggs in the stem below the cotyledon. In the lab, oviposition started on the third day after adult emergence (Talekar and Lee, 1988). Larvae (Fig. 2) emerge from eggs and feed on plant cortex under the stem epidermis (CPC Report, 1997). The duration of the larval period (at 28°C) is 9 to 14 days (10.88 days on average). Considerable mortality has been observed in the egg and larval stages. Up to 13 eggs per plant may be oviposited by *O. centrosematis*, but an average of two larvae per plant reach the pupation stage. Pupation takes place just below the epidermis at the root-shoot junction in the plant cortex. Pupation ranged from 10 to 13 days in the laboratory (Talekar and Lee, 1988).

Adults emerge during the day (CPC Report, 1997). Temperature appears to play a key role in controlling adult emergence. In the lab, adult males lived 6 to 24 days (15 days on average) and adult females lived 6 to 21 days (12 days on average) (Talekar and Lee, 1988). Mating tends to occur between 0500 and 0800 hours. The pre-mating period lasts 2.5 days, pre-oviposition 3.5 days and oviposition 12.2 days. Characteristic feeding scars are found in hypocotyls where oviposition by *O. centrosematis* occurs. Other agromyzid flies (e.g., *O. phaseoli*

and *Melangromyza sojae*) also make feeding punctures on foliage (Talekar and Lee, 1988).

Pest Importance

O. centrosematis is a minor pest of most legumes in Asia and Africa. Damage sometimes goes unnoticed in the presence of the more dominant *O. phaseoli*. In Uttar Pradesh, India, however, *O. centrosematis* is a destructive pest of peas (*Pisum sativum*). Singh et al. (1981) found that more than 95% of the damaged plants die when the crop is planted during the first week of October. Plant mortality is reduced in crops planted in November.

Centrosema pubescens in Java and Malaysia, and *Calopogonium mucunoides* in Malaysia, are important cover plants for rubber plantations, which may be effectively destroyed by *O. centrosematis* (Spencer, 1973).

On soybean, *O. centrosematis* can be responsible for considerable yield loss and, at times, high mortality (Talekar and Lee, 1988).

Symptoms/Signs

Adults are active during the day and feed exclusively on legume plants. Larvae feed inside the stem cortex, below the cotyledons, resulting in destruction of the cortex tissue and an accumulation of frass. Larvae can be seen feeding in tunnels inside the stem when the stem is split open. Pupae are found in the same layer but at the root-shoot junction. In severe cases, the plant looks wilted and eventually dies. Adults make oviposition and feeding punctures in the hypocotyl, but these punctures are very small and barely seen by the naked eye. Plants are often yellow and stunted. Stems are often thicker than normal and cracked lengthwise just above the soil line. In cases of heavy infestation, many plants may die.

Known Hosts

O. centrosematis appears to be restricted to the legumes (Fabaceae) (AVRDC Crop Protection Guides, 2001).

Major Host:

Glycine max (soybean), *Centrosema pubescens* (Centro), *Crotalaria pallida* (smooth crotalaria), *Phaseolus vulgaris* (common bean), and *Pisum sativum* (pea) (CPC Report, 1997).

Minor Hosts:

Cajanus cajan (pigeon pea), *Calopogonium*, *Lablab purpureus* (hyacinth bean), *Macrotyloma uniflorum* (horsegram), *Medicago sativa* (alfalfa), *Phaseolus lunatus* (lima bean), *Pueraria*, *Tephrosia candida* (hoang pea), *Vigna angularis* (adzuki bean), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), and *Vigna unguiculata* (cowpea) (CPC Report, 1997).

O. centrosematis completed a full life cycle on the following plants in a laboratory setting: soybean, mungbean, snapbean, adzuki bean, cowpea, pigeonpea, horsegram, and alfalfa (Talekar and Lee, 1988).

Known Distribution

O. centrosematis is localized in China, Australia and India. It is widespread in Taiwan and Indonesia. It is reported as present in Japan, Malaysia, Thailand, Kenya, Malawi, Tanzania and Uganda.

Potential Distribution Within the US

Information is not available at this time.

Survey

All beanflies prefer to feed on young plants. Larvae feed in plant cortex just under stem epidermis. Since feeding is internal, cut the main stem just above the soil line, open and look for mining and the small white maggot (larva) (AVRDC Crop Protection Guides, 2001). Damage to soybean by *O. centrosematis* is often confused with that of *O. phaseoli*, another cortex-feeding agromyzid fly often co-occurring with *O. centrosematis* (Talekar and Lee, 1988; Talekar and Huang, 1993).

Key Diagnostics

The larva is a small white maggot with a brown head. The adult is a tiny black fly with transparent wings, about ¼ the size of a common housefly (AVRDC Crop Protection Guides, 2001).

Ophiomyia phaseoli

Scientific Name
Ophiomyia phaseoli Tryon

Synonyms:
Agromyza destructor, Agromyza fabalis, *Agromyza phaseoli*, *Melanagromyza phaseoli, Melanagromyza similes, Oscinis fabae, Oscinis phaseoli*

Common Name(s)
Bean fly, agromyzid fly, bean stem maggot, french bean fly, french bean miner, katjang fly, legume root miner, pea stem agromyza, pea stem fly, pea stemborer, snapbean fly, soybean miner, stemborer

Type of Pest
Fly

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family**: Agromyzidae

Reason for Inclusion in Manual

Pest Description
The egg is oval, milky white, opaque or translucent measuring 0.30 to 0.39 mm long and 0.10 to 0.17 mm wide.

O. phaseoli larvae are cortex feeders and pupate in the cortex, usually at the root-shoot junction. Pupae can often be seen sticking under the membranous epidermis. In all host plants, oviposition takes place in unifoliate or early trifoliate leaves.

Figure 1. Adult *O. phaseoli*, male (left) and female (right). Photo by James Litsinger (CABI, 2004)

O. phaseoli pupae are barrel-shaped. The cephalic end is somewhat pointed and the posterior end is slightly rounded. There are twelve visible segments. When newly emerged, pupae are yellow with a brownish tinge and with distinctly darker ends. The segments are well defined and the anterior and posterior spiracles are black. Shortly before adults emerge, the puparia become dark brown. Puparia measure from 2.02 to 2.30 mm long and 0.81 to 1.05 mm wide (CABI, 2004).

Average adult fly measurements are 2.07 mm long and 4.97 mm wide, including wing expanse. Females are slightly bigger than males (Fig. 1). Females are 1.88 to 2.16 mm long and 0.70 mm wide at the thorax, with a wing expanse of 4.45 mm. In males, the body is 1.60 to 1.84 mm long and 0.60 mm wide at the thorax, with a wing expanse of 3.80 mm.

Biology and Ecology:

Fertilized *O. phaseoli* females are most active on warm clear days. They are active fliers and seek tender leaves on the host plant for oviposition. Adults show a distinct preference for younger legume hosts for oviposition and feeding. They tend to lay eggs during the morning hours on the upper side of the leaves, often near the midrib close to the petiole. The eggs are inserted between the epidermis and spongy parenchyma. In all legume host plants, *O. phaseoli* lays eggs in the leaves, especially the unifoliate leaves. However, a biotype of this species found on soybean in Indonesia oviposits in cotyledons in addition to unifoliate leaves (CABI, 2004).

Between 10 and 15% of leaf punctures contain eggs; remaining punctures are made by adults while feeding on plant sap. The number of eggs laid per female varies considerably. In common bean (*Phaseolus vulgaris*), the females lay a lifetime average of 94 eggs (range 16 to 183). Females lay a lifetime average of 77 eggs in cowpea. The oviposition period begins 3 to 4 days after adult emergence and continues for 10 to 15 days. The incubation period of the eggs varies from 2 to 4 days, depending upon temperature.

The eggs may hatch at any time of the day. *O. phaseoli* undergoes three larval instars. The newly hatched, pale, yellowish-white first-instar larvae remain motionless for 1 to 2 hours before feeding. The first instar feeds mainly in leaf blade tissue before entering the midrib and eventually entering the stem. The first larval stadium lasts from 1.7 to 2 days with a mean of 1.9 days. Second-instar larvae initially still feed inside the midrib but soon enter the petiole and usually molt into the third instar at the petiole stem junction. The duration of second instar lasts from 2 to 2.4 days. The third instar feeds voraciously in the stem just beneath the epidermis. In young seedlings, the larvae may feed as low as the roots, but in most cases feeding only extends to just below the soil surface. The duration of the third instar varies from 4.5 to 5.5 days (mean of 4.7 days) in common bean and 3 to 4 days (mean of 3.33 days) in cowpea. Before pupation, the fully grown larvae cease feeding for 6 to 10 hours and construct a semicircular hole in the epidermis for the emerging adults to escape from the

pupae. The prepupal period lasts 1.5 to 2 hours. The freshly formed pupae become opaque (CABI, 2004).

The site of pupation varies depending upon the stage and condition of the host plant. During the seedling stage, pupation normally takes place beneath the epidermis of the stem, near the soil surface. In the later stages of the host plant, pupation can take place at the junction of the leaf lamina and petiole. In some instances, pupation is observed in the midrib of the leaflet. Pupal period varies according to temperature. It is reported that below 22 °C, the pupation period ranged from 11 to 14 days. At 28°C, the prepupal period shortened to 10 to 12 days, and at 32°C, the pupal period lasted 8 to 9 days.

Fully developed adults emerge from the puparia via a transverse T-shaped slit or a crack made by the ptilinum. Immediately after emerging, the soft bodied and unpigmented adults remain motionless while the wings unfold and the exoskeleton hardens and darkens. The adults attain a metallic black color after about an hour. The adults fly about 4 to 5 hours after emergence.

Adult females live for 23 to 42 days and males for 31 to 38 days under undefined laboratory conditions. If no food is provided, they die in 2 to 3 days, while a second study reported a much shorter adult longevity; 7.13 ± 2.39 and 15.42 ± 3.78 days, respectively, for males and females under laboratory conditions. The life span averaged 49 hours for starved flies, 94 hours for flies provided with water only, and 212 hours for flies provided with glucose solution (CABI, 2004).

Adults feed on three general food sources: water droplets on the leaves, natural secretions of plants, and host-plant sap exuding from feeding and ovipositional punctures made on the leaves. Adults start mating after an average pre-mating period of 18 hours. Copulation takes place only during the day and lasts from 4 to 94 minutes, with an average of 18.5 minutes under laboratory conditions. Duration of copulation is between 1 and 2 hours, usually occurring on the upper surface of the leaves. Males and females mate several times during their life.

In Java, Indonesia, the maximum number of generations of *O. phaseoli* per year is 14, while in the Philippines and in Australia there are between 9 and 11 generations per year. In India, 8 to 9 generations occur between July and the following April and in Egypt, there were 10 to 12 generations per year (CABI, 2004).

Life-table studies found that in common bean (*Phaseolus vulgaris*) the differences in the initial number of eggs laid was not great among four or five observations, but that the total survival rate from eggs to adults was season dependent. The survival rate was much higher in summer (14 to 20%) than in winter (3 to 6%) or spring (8%). The location of puparia within the host plant during different seasons appeared to cause this variation. In summer, pupation takes place in the lower part of the stem or beneath the soil; in winter it occurs in

the upper part of the stem. Such seasonal changes in the site of pupation affect parasitism by pupal parasites, especially Pteromalids; summer pupae suffer much less from pupal parasitism.

Pest Importance

In tropical to subtropical Asia, *O. phaseoli* remains a destructive pest of most food legumes, particularly common bean, cowpea, mungbean, blackgram, lima bean and soybean (at least in Indonesia). The nature of the extent of *O. phaseoli* damage in different hosts varies from crop to crop and season to season. In general, however, plants are more seriously damaged in the seedling stage than later stages. The consequences of insect attack in the seedling stage, if the plant survives, are manifested in the older plants. In general, the yield during the rainy season is much less than in the dry season. In Java, Indonesia, in 30 observations at Bogor, up to 100% of common bean plants were damaged, with high plant mortality and yield loss. In Tanzania, the yield loss ranges from 30 to 50%. In New South Wales, Australia, it was impossible to grow common bean, indicating thereby 100% plant damage and total yield loss if plants are not adequately protected. In Taiwan, *O. phaseoli* causes 35% yield loss in common bean (CABI, 2004).

In Indonesia, a biotype of *O. phaseoli* attacks soybean. Whereas in the rest of its range, *O. phaseoli* lays eggs in the leaves of host plants, in Indonesia the biotype lays eggs in soybean cotyledons soon after these plant parts emerge above ground. Larvae, after initial feeding in cotyledons, enter the stems and in most cases kill the soybean plant. The extent of damage and subsequent yield loss varies from season to season. In dry seasons (June to October) the plant mortality can be 80%, compared to 13% in the wet season (November to April). *O. phaseoli* causes very little if any loss in soybean crops in the rest of its range.

Symptoms/Signs

The most serious damage by adults occurs when plants are at the unifoliate stage. The unifoliate leaves show a large number of feeding and oviposition punctures on the upper side with corresponding light yellow spots, especially on the basal portion of the leaf. The first and second trifoliate leaves show some egg holes, but leaves situated above this are practically undamaged. Larval feeding soon after hatching produces

Figure 2. Cowpea foliage with severe damage, infested leaves become blotchy and later hang down. Photo courtesy of CABI, 2004.

numerous larval mines, which are better seen on the underside of the leaves just under the epidermis and appear as silvery, curved stripes. On the upper side of the leaf, only a few tunnels are visible. Later, both egg holes and larval mines turn dark brown and are clearly visible. In cases of severe attack, infested leaves become blotchy and later hang down (Fig. 2). These leaves may dry out and may even be shed. When mature plants become infested, insect damage is confined to the leaf petioles, which become swollen and at times the leaves may wilt (CABI, 2004).

The developing second and third instar larvae mine downward into the cortex just underneath the epidermis. The third instar continues to feed downwards into the tap root and returns to pupate still inside the stem, close to the soil surface. The feeding tunnels are clearly visible on the stems (Talekar, 1990). If the *O. phaseoli* larvae population is high, larval feeding leads to destruction of the cortex tissue around the root-shoot junction. This initially leads to yellowing of the leaves, stunting of plant growth and even plant mortality. If the damage is less severe, the root-shoot junction area appears swollen. In some cases, the host plant produces adventitious roots above this swollen area on the stem.

In Indonesia, where a biotype of *O. phaseoli* attacks soybeans soon after emergence, larval tunnels in cotyledons are clearly visible (Talekar, 1990). Later, damaged cotyledons turn yellow and are shed. In most cases, the plant is killed within 10 to 15 days of emergence.

Known Hosts

Major Hosts

Fabaceae (leguminous plants), *Phaseolus (beans)*, *Phaseolus vulgaris* (common bean), and *Vigna radiata* (mung bean)

Minor Hosts

Cajanus cajan (pigeon pea), *Crotalaria juncea* (sunn hemp), *Crotalaria pallida* (smooth crotalaria), *Glycine max* (soybean), *Lablab purpureus* (hyacinth bean), *Macrotyloma uniflorum* (horsegram), *Medicago sativa* (alfalfa), *Phaseolus lunatus* (lima bean), *Pisum sativum* (pea), *Psophocarpus tetragonolobus* (winged bean), *Vigna aconitifolia* (moth beans), *Vigna angularis* (adzuki bean), *Vigna mungo* (black gram), *Vigna sinensis* ssp. *sesquipedalis* (asparagus bean), and *Vigna unguiculata* (cowpea)

Wild hosts

Cyamopsis tetragonoloba (clusterbean), *Mucuna pruriens* (Buffalobean), *Phaseolus coccineus* (runner bean), and *Phaseolus lathyroides* (Phasey bean)

Known Distribution

Present in 50 countries on all seven continents including Bangladesh, Brunei Darussalam, China, India, Indonesia, Iran, Israel, Japan, Jordan, Laos, Malaysia, Myanmar, Nepal, Pakistan, Philippines, Saudi Arabia, Singapore, Sri Lanka,

Thailand, Vietnam, Burundi, Congo Democratic Republic, Egypt, Ethiopia, Kenya, Libya, Madagascar, Malawi, Mali, Mauritius, Nigeria, Reunion, Rwanda, Senegal, South Africa, Sudan, Tanzania, Uganda, Zambia, Zimbabwe, U.S., Australia, Belau, Federated States of Micronesia, Fiji, Guam, Northern Mariana Islands, Papua New Guinea, Samoa, and the Solomon Islands (CABI, 2004)

Potential Distribution Within the US

Present in Hawaii but not reported in the continental U.S.

Survey

Adults of *O. phaseoli* are extremely agile and will fly when slightly disturbed. They remain stationary only while laying eggs, usually at the basal portion of the leaves. Eggs are confined to unifoliate and early trifoliate leaves. Eggs of one biotype in Indonesia can be found in cotyledons, but only on soybean plants. Upon cutting the stem open, a brown or dark feeding area of damaged tissue can be seen just underneath the epidermis. This portion will contain both larvae and pupae (CABI, 2004).

Key Diagnostics

Identification of adults of *O. phaseoli* in the field is difficult, because they do not cause significant damage. Adults are agile and thus difficult to observe in the field and, to an inexperienced person, they can be easily confused with adults of other agromyzid species. At least two other species of agromyzids, *O. centrosematis* and *Melanagromyza sojae*, attack most economically important legumes simultaneously with *O. phaseoli*. For practical purposes, it is much easier to identify *O. phaseoli* and other agromyzids by observing larvae and pupae. Besides morphological differences, their feeding and oviposition sites within the host plants give a fairly accurate idea of their identity (CABI, 2004).

Both larvae and pupae can be identified by observing their spiracles. In both stages anterior spiracles are small, with a circle of six minute bulbs. Posterior spiracles closely adjoin on the conical projections, usually with about 10 minute bulbs. Puparia are pale yellow, straw colored or light brown.

Rivellia quadrifasciata

Scientific Name
Rivellia quadrifasciata Macquart

Common Name(s)
Soybean nodule fly

Type of Pest
Fly

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Platystomatidae

Reason for Inclusion in the Manual

Pest Description
Adults (Fig. 1) are small clear-winged flies (3.6 to 6.4 mm long), with four transverse black bands across each wing, oval reddish brown head, dark brown or black thorax, and rusty red abdomen. Eggs are cylindrical with pointed ends, chalky to creamy white in color, and 1.0 x 0.3 mm in size. Larvae (Fig. 2A) are small (8.0 x 1.4 mm) white maggots that live in the soil. The pupae (Fig. 2B) are russet to mahogany in color, and 4.8 x 1.6 mm in size.

Figure 1. Adult fly of *Rivellia quadrifasciata*. Photo courtesy of J. W. Van Duyn.

Biology and Ecology
Larvae of *R. quadrifasciata* overwinter as a last-instar in the soil beneath soybean stubble. Most larvae are found 10.2 to 15.2 cm below the soil surface (Koethe, et al., 1986). Larvae can move vertically in the soil, but their horizontal displacement is minimal. Pupae

develop as temperature increases in the spring. Adult emergence begins in May and continues into July, but they peak in late June and early July (Koethe, et al., 1986). Generally, adults of *R*. *quadrifasciata* are present in soybean fields from May through October. Adults are diurnal and mating can take place on larval host plants, as well as on non- host plants. Eggs are oviposited in the soil and in crop residue around the bases of potential larval host plants. Most eggs are oviposited within 2 mm of the soil surface, but they can be found in deeper zones if females enter soil cracks and crevices (Koethe and Van Duyn, 1988).

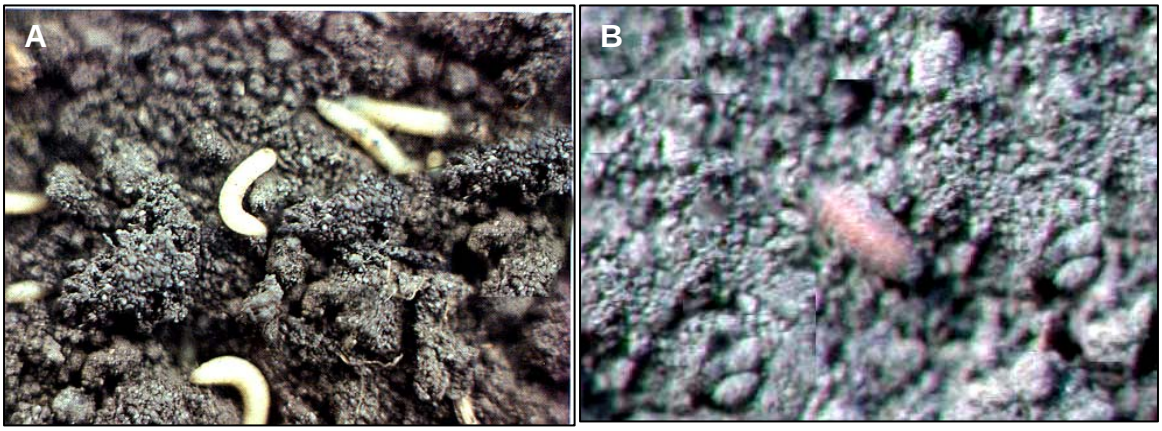


Figure 2. Larvae (A) and Pupae (B) of *Rivellia quadrifasciata*. Photos courtesy of J. W. Van Duyn.

Adults of *R*. *quadrifasciata* feed on a variety of foods, including carrion, honeydew, bird droppings, insect frass, nectar and other carbohydrate sources. Females are mostly attracted to proteinaceous food sources, such as, carrion, meat and dead insects, while both males and females are attracted to honeydew and nectar (Koethe and Van Duyn, 1989). There are no control strategies for soybean nodule fly available, because it is not thought to significantly impact soybeans yields.

Pest Importance

The soybean nodule fly is a fairly common insect. Its potential importance as an economic pest was realized only when it was discovered that the larvae feed on soybean nodules (Eastman and Wuensche, 1977). In studies with potted plants, nodule damage by chewing insects has shown to stimulate root growth and branching, as well as reduced seed yield. Presumably, high populations of nodule fly maggots could reduce yield, especially under high yield conditions where the demand by plants for nitrogen is great. Severe infestations may produce symptoms of nitrogen deficiency (Van Duyn, 2004).

Symptoms

Symptoms caused by *R*. *quadrifasciata* are not easily visible. All damage occurs below ground. Upon hatching, larvae feed on and destroy the nitrogen fixing nodules of the soybean plant; damaged nodules appear hollowed-out (Van Duyn,

2004). Studies with *R*. *quadrifasciata* and other *Rivellia* species suggest that nodules may be essential for larval development.

Known Hosts

Larvae of flies of the genus *Rivellia* are known to feed on the nitrogen-fixing root nodules of legumes including: soybean, peanut, beans, peas, and other cultivated and wild beans (Koethe and Van Duyn, 1984). Several species, notably *R*. *quadrifasciata*, have been reported to be in transition from wild to cultivated plants and have acquired importance as pests of soybean, Southern pea, and *Vigna unguiculata*, in the southeastern U.S.

Known Distribution

Rivellia quadrifasciata is a New World species. It is reported to be common in the eastern states of the U.S. (Namba, 1956). Its distribution in all the major soybean-producing states needs to be documented.

Survey

Adults of *R*. *quadrifasciata* can be monitored by using traps. These include: sticky-traps, pitfall traps, sweeping nets, and others (Koethe, et al., 1986; Koethe and Van Duyn 1989). Koethe and Van Duyn (1989) reported that fruit, ethylene glycol and soap water baits caught few flies unless meat was added. Traps baited with meat or dead insects trapped only females. Vertically placed meat-baited jar traps and sticky traps showed a contrasting distribution of both males and females within and above the soybean canopy, but catches of females were not influenced by male distribution. Females were active near the soil surface, whereas males tended to concentrate within or above the plant canopy. Sticky traps caught both males and females and appeared to be as effective as bait traps for females.

Key Diagnostics

Adults of soybean nodule fly (Fig. 1) can be found flying in the canopy of soybean plants. They are small clear-winged flies (3.6 to 6.4 mm long), and typically have four transverse black bands across each wing, rusty red abdomen, oval reddish brown head, and dark brown or black thorax.

Mealybugs

Maconellicoccus hirsutus

Scientific Name
Maconellicoccus hirsutus Green

Synonyms:
Maconellicoccus pasaniae, Maconellicoccus perforatus, Paracoccus pasaniae, Phenacoccus glomeratus, Phenacoccus hirsutus, Phenacoccus quaternus Pseudococcus hibisc, Spilococcus perforatus

Common Name(s)
Pink hibiscus mealybug, hibiscus mealybug, hirsutus mealybug, pink mealybug

Type of Pest
Mealybug

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Pseudococcidae

Reason for Inclusion in Manual

Pest Description
Eggs: Freshly laid eggs of the pink hibiscus mealybug (PHM), M*aconellicoccus hirsutus*, are orange but become pink before they hatch.

Larvae: Crawlers (0.3 mm long) are pink. Immature females and newly matured females have grayish-pink bodies dusted with mealy white wax (Fig. 2)

Adult Females: The adult female is 2.5 to 4 mm long, soft-bodied, elongate oval and slightly flattened (Fig. 2); on maturation, she

Figure 1. Eggs of *M. hirsutus*. Photo courtesy of Marshall Johnson, Department of Entomology, University of Hawaii at Manoa.

begins to secrete sticky, elastic, white wax filaments from her abdomen to form a protective ovisac for her eggs. As her pinkish-grey body fills with salmon-pink eggs, it assumes a pink color, but this is often not immediately visible because the entire colony tends to become covered by white, waxy ovisac material. When the sticky ovisac wax is parted with a needle, clusters of pink eggs and pink to grey females become visible. On microscopic examination of slide-mounted females, the combination of 9-segmented antennae, anal lobe bars, numerous dorsal oral rim ducts on all parts of the body except the limbs and long, flagellate dorsal setae make the species fairly easy to recognize in parts of the world where other *Maconellicoccus* species do not occur (CABI, 2004).

Adult Males: Males have one pair of very simple wings, long antennae, white wax filaments projecting posteriorly (Fig. 3) and lack mouthparts.

Biology and Ecology

Local movement of *M. hirsutus* occurs at the first instar (crawler) stage. Crawlers are very small (0.3 mm long), light, and can survive a day or so without feeding. They cannot walk far by themselves, but are ideally suited to transport by water, wind and animal agents, including domestic animals and man. Misra (1920) recorded transport of *M. hirsutus* by nymphs of another mealybug species (*Ferrisia virgata*) in India. Accidental introductions to new countries apparently occur via infested plant material.

Figure 2. Various stages in the life cycle of the pink hibiscus mealybug (adult female (arrow) and offspring. Photograph courtesy of Dale Meyerdirk, APHIS.

Once the crawler settles at a feeding site, development continues; there are three instars in the female and four in the male. Crawlers settle in cracks and crevices, usually on new growth, which becomes severely stunted and distorted. Densely packed colonies develop in these areas; Kairo et al. (2000) describe the symptoms in detail. Reproduction is reported as mostly parthenogenetic in Egypt and Bihar, India (CABI, 2004) In West Bengal, India, *M. hirsutus* is recorded as being biparental and it seems likely that populations in the West Indies are also biparental (Williams, 1996). Males are reported to have a pupal stage capable of locomotion (Bartlett, 1978).

Figure 3. Adult male pink hibiscus mealybug, Notice the two long waxy ‘tails’. Photo courtesy of Marshall Johnson, Department of Entomology, University of Hawaii at Manoa.

The life cycle has been studied in India (Mani, 1989). Each adult female lays 150 to 600 eggs over a period of about one week, and these

hatch in 6 to 9 days (Bartlett, 1978; Mani, 1989). A generation is completed in about five weeks in warm conditions (Bartlett, 1978), although there are reports of a generation time of as little as 23 days in the laboratory. In countries with a cool winter, the species survives cold conditions as eggs (Bartlett, 1978) or other stages, both on the host plant and in the soil (Pollard, 1995). There may be as many as 15 generations per year (Pollard, 1995).

Infestations of *M. hirsutus* are often associated with attendant ants (Ghose, 1970), which collect sugary honeydew from the mealybugs. Ants recorded attending *M. hirsutus* include *Oecophylla* spp., *Iridomyrmex* spp. and *Solenopsis* spp. (Williams and Watson, 1998) in the Solomon Islands and Papua New Guinea.

Pest Importance

Williams (1996) summarizes records of damage caused by *M. hirsutus*. Almost all serious damage by the mealybug has been recorded between 7° and 30° north latitude, where there are reports of seasonal differences in the incidence of the pest. Direct feeding on young growth (stems, leaves and flowers) causes severe stunting and distortion including crinkling of the leaves, thickening of stems and a bunchy-top appearance of shoots; in severe cases the leaves may fall. Honeydew and sooty mold contamination of fruit may reduce their value (Garland, 1998). In India, stunted and distorted growth caused by *M. hirsutus* in mulberry is known as Tukra disease (Rao et al., 1993) and is a problem in most of the silk producing areas (Tewari et al., 1994). It has been suggested that symptoms associated with *M. hirsutus* infestation may be due to a virus infection on cacao in Zanzibar (de Lotto, 1967) and on mulberry in India (Tewari et al., 1994).

Francois (1996) estimated annual losses in Grenada due to *M. hirsutus* damage to crops and environment at US $3.5 million before biological control agents were established. In the first few years of the mealybug problem in the Caribbean islands, affected countries suffered serious loss of trade, because other countries would not accept shipments of agricultural products from them (Peters and Watson, 1999). In the period 1995 to 1998, Peters (1999) estimated the island's overall losses and costs at US $8.3 million, of which the control program cost US $1.1 million (Kairo et al., 2000). Overall losses and costs to St. Kitts in 1995 to 1997 were estimated by Francis (1999) at US $280,000, with an additional loss of trade estimated at US $22,000. For St. Lucia, losses were estimated at US $67,000 (Anon., 1999), and for St. Vincent and the Grenadines losses were estimated at US $3.4 million (Edwards, 1999). If the mealybug were to spread across the southern U.S., it is estimated that it could cause losses of US $750 million per year (Moffit, 1999).

Other crops seriously damaged by *M. hirsutus* include cotton in Egypt, with growth sometimes virtually halted (Hall, 1921); tree and herbaceum cotton in India, with reduction in yield (Dhawan et al., 1980; Muralidharan and Badaya,

2000); the fiber crops *Hibiscus sabdariffa* var. *altissima* (roselle), *H. cannabinus* (mesta) and *Boehemeria nivea* in West Bengal, India, and Bangladesh (Ghose, 1961, 1972b; Singh and Ghosh, 1970), with reduction in fiber yield of roselle; grapes in India, with up to 90% of bunches destroyed in the Bangalore area (Manjunath, 1985) and heavily infested bunches made unfit for consumption or marketing (Vereesh, 1986); pigeonpea in India (Patel et al., 1990); *Zizyphus mauritiana* in India (Balikai and Bagali, 2000); ornamental Hibiscus in Papua New Guinea (Williams and Watson, 1988); and cacao in the Solomon Islands (Williams and Watson, 1988) and Grenada (Pollard, 1995).

Experimental evidence suggests that Tukra-diseased leaves may be more nutritious to silkworms than normal leaves (Ahamed et al., 1999).

In Grenada, severe devastation of natural habitats was seen, for example, in the Grand Etang area where a stand of 38 ha of blue mahoe (*Hibiscus elatus*) was destroyed (Peters and Watson, 1999; Kairo et al., 2000). This tree is dominant in the natural rainforest; if such devastation had become widespread, the watersheds and soils of the island would have been threatened (CABI, 2004).

In Grenada, where the infestation remained unchecked for over a year, the mealybug extensively devastated amenity plantings and landscaped gardens in hotels, resulting in serious losses to the tourist industry and people employed therein. In addition, cash crops also produced little or no return for 1 to 2 years, which impacted farming income and agricultural trade (Peters and Watson, 1999). Such damage has a major impact on small island economies.

Symptoms/Signs

The saliva that *M. hirsutus* injects into the host plant while feeding probably contains a substance that is phytotoxic (Williams, 1996). Host-plants differ in their susceptibility to the toxin (Fig. 4, 5, 6). The more tolerant species tend to be infested at their growing points and in stem axils and infested new growth

Figure 4. Hibiscus twig (left) and shrub (right) both heavily infested by pink hibiscus mealybug. Photo courtesy of Dale Meyerdirk, APHIS

becomes stunted, with reduced internode extension and leaf expansion. Stunted stems may become swollen. In more sensitive plants, stunting is more marked and new growth forms cabbage-like clusters ('bunchy tops', with the mealybugs hidden in the creases of the growth (Fig. 5). In highly susceptible plants, even brief probing of unexpanded leaves by crawlers causes severe crumpling of the leaves when they subsequently expand, while established infestation can cause total defoliation and even death of the whole plant. As the plant dies back from the tips, the mealybugs migrate to healthy tissue, moving from shoot tips to twigs to branches and finally down the trunk. In heavier infestations, white masses of wax concealing mealybugs may occur in axils and on twigs and stems (Fig. 4, 6) (CABI, 2004). *Samanea saman* is severely affected. Plant tissues are often coated with sooty mold.

It should be noted that the mealybug *Paracoccus marginatus* causes very similar damage on Hibiscus to that caused by *M. hirsutus* (Pollard, 1999).

Known Hosts

M. hirsutus is highly polyphagous and has been recorded feeding on hosts from 76 plant families (Ben-Dov and German, 2003) and over 200 plant genera (Levy, 1996); it shows some preference for hosts in the families Malvaceae, Leguminosae and Moraceae. Mani (1989), Garland (1998), Miller et al. (1998) and Ben-Dov and German (2003) give extensive host lists. When introduced to tropical countries in the absence of any natural enemies, *M. hirsutus* attacks a wide range of (usually woody) plants including agricultural, horticultural and forest species. It has been recorded attacking cotton and soybean, both annuals that are rarely attacked by mealybugs (Williams, 1986). However, in the Caribbean it has only developed seriously damaging populations on fewer than 20 host-plant species (Kairo et al., 2000). If *M. hirsutus* spreads into the southern U.S. and southern Europe, it could threaten crops like grapes and cotton (Williams, 1996). One of the most favored hosts is *Hibiscus rosa-sinensis*. *M. hirsutus* can be reared in the laboratory on pumpkins, particularly those varieties with creases in the skin (Japanese pumpkin, *Cucurbita moschata*; acorn squash, *Cucurbita pepo* var. *turbinata*) and on sprouting Irish potatoes (Mani, 1990; Meyerdirk, 1997; Serrano and Lapointe, 2002).

Figure 5. 'Bunchy top' damage. Photo courtesy of Ru Nguyen.

Major hosts
Abelmoschus esculentus (okra), *Allamanda, Alpinia purpurata* (gingerlily), *Annona* spp., Artocarpus (breadfruit trees), *Averrhoa carambola* (carambola), *Boehmeria nivea* (ramie), *Bougainvillea, Cajanus cajan* (pigeon pea), Citrus, *Glycine max* (soybean), *Gossypium spp.* (cotton), *Hibiscus* spp., *Malpighia glabra* (acerola), *Manilkara zapota* (sapodilla), *Morus* spp. (mulberry), *Morus alba* (mora), *Musa x paradisiaca* (plantain), *Passiflora edulis* (passionfruit), *Persea americana* (avocado), *Samanea saman* (rain tree), *Sida acuta* (Sida), *Spondias* (purple mombin), *Spondias purpurea, Tectona grandis* (teak), *Theobroma cacao* (cocoa), and *Vitis vinifera* (grape).

Figure 6. *Maconellicoccus hirsutus* on Florida triema. Photo courtesy of Ru Nguyen.

M. hirsutus forms dense colonies in cracks and crevices. The severe distortion of new growth caused by the mealybug on many hosts, creates a microhabitat for them (Ghose, 1972a; Beardsley, 1985). These colonies can be difficult or impossible for natural enemies to reach, especially coccinellid predators (CABI, 2004).

Known Distribution
M. hirsutus is probably native to southern Asia (Williams, 1996) and has been accidentally introduced to other parts of the world. The pest is widespread in Asia, Africa, and in Central America. The most recent introductions were in North America (California, Florida and Mexico) and the Caribbean. It has spread to more than 25 territories and is still extending its range (Kairo et al., 2000). It occurs as far north as Lebanon, so there is no reason why it should not be able to colonize the southern U.S., southern Europe and parts of the Middle East where it is not yet known to occur (CABI, 2004).

Potential Distribution Within the US
The pest is present in California, Florida and Mexico. There is no reason why it should not be able to spread and colonize states in the southern U.S. (CABI, 2004).

Survey
Examine plant material, especially growing tips, for distorted, stunted, bunchy growths containing white woolly wax, tiny salmon-pink eggs, and sooty mold or sticky honeydew. The honeydew produced may attract attendant ants. The entire mealybug colony tends to become covered by white, sticky, elastic, woolly, wax

ovisac material. When the sticky ovisac wax is parted with a needle, clusters of pink eggs and pink to grey females become visible. In heavier infestations, white masses of wax concealing mealybugs may occur in axils and on twigs and stems. Good light conditions are essential for examination; in poor light, a powerful flashlight is helpful. One of the most common and favored hosts of *M. hirsutus* is *Hibiscus rosa-sinensis*; this is a good host to monitor for early detection of the arrival of the pest (CABI, 2004).

Key Diagnostics

In parts of the world where other species of *Maconellicoccus* do not occur, slide-mounted adult females of *M. hirsutus* are fairly easy to recognize. Examination of slide-mounted material is advisable because some other species of mealybug are similar to *M. hirsutus* in appearance and damage caused, for example, *Phenacoccus solenopsis* and *Paracoccus marginatus*. *P. marginatus* differs from *M. hirsutus* in the field by having yellow body contents, not pink. When preserved in 80% alcohol, specimens of *P. marginatus* turn black in a matter of days, whereas *M. hirsutus* remain brown.

Mites

Eutetranychus africanus

Scientific Name
Eutetranychus africanus Tucker

Synonyms:
Anychus africanus, Eutetranychus cendani, Eutetranychus sambiranensis,

Common Name(s)
African red mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

Reason for Inclusion in Manual

Pest Description
The body of the female mites is 0.3 to 0.4 mm long and 0.3 mm wide, dark red or brown close to black in color. It has a round to oval shape. The male is light brown and slightly smaller than the female. The front side is widest and the back side rather pointed. Body length is about 0.3 to 0.4 mm and the width about 0.3 mm. Dorsocentral setae short, spatulate to subspatulate; tibia II with 6 setae; coxa II with 2 setae; spermatheca rounded.

Figure 1. Adult stages of *Eutetranychus africanus*. Photo courtesy of 1704Hhttp://www.ipmthailand.org/en/Pests/African_red_mite.htm

Biology and Ecology
Mating starts as the male finishes

molting and becomes an adult. The male will start looking for a third instar larva female, and once found will wait for the final molting of the female. Mating then takes place immediately, and 1 to 2 days after mating the female will start to produce eggs.

The development of both the males and females takes about 9 days, including about 5 days for the egg stage and around 1.5 days for each of the 3 larval instars. Males live only for 1.5 days and females live for approximately 8 days, during which time the females lay on average 14 eggs.

Pest Importance

E. africanus was the most important pest mite on *Ziziphus mauritiana* in Thailand. *E. africanus* also affects growth of papaya in Thailand if not controlled (Childers, 2006).

Symptoms/Signs

The African red mite sucks leaf phloem at the upper surface of leaves. The pest multiplies rapidly during hot and dry weather conditions. The infested leaves show whitish spots where the mites have fed. Heavy infestations produce numerous fine stippled areas on the leaves causing them to drop prematurely without turning brown. The leaves become pale and lack a glossy green color like normal leaves. The trees can tolerate quite a number of mites, but heavy infestations may result in leaf shedding, which will affect the development of flowers and fruits.

Known Hosts

The African red mite can be found on a wide range of hosts including: durian, papaya, tangerine, pomelo, lime, leech lime, *Citrus sinensis*, jackfruit, breadfruit, horse-radish, cassava, cotton, soybean, cowpea, castor bean, watermelon, garden pea, and hibiscus.

Known Distribution

The African red mite is an important pest of durian in Thailand, especially during the cool season (late October to early March). The pest has also been reported from Burma, Comoros, Egypt, India, Madagascar, Mauritius, Mozambique, Papua New Guinea, Reunion, and South Africa.

Potential Distribution Within the US

Information is not available at this time.

Survey

Mites are small and are hardly visible to the naked eye appearing as small reddish or brown dots on the leaf surface. An easy way to observe them is shaking an infested leaf above a sheet of white paper. Use a small hand lens to

observe their behavior on the leaves. Be sure to check leaves that have the stippling effect of mite feeding.

Key Diagnostics

The small size of mites makes it difficult for farmers or field workers to recognize them. Inexperienced observers may easily confuse mites with other small animals that live on the leaves, such as thrips. Correct identification is also important, because not all mites are pests. Several predatory mites can be found on the leaves as well, and these are beneficial as natural enemies of the red mites.

Eutetranychus orientalis

Scientific Name
Eutetranychus orientalis Klein

Common Name(s)
Citrus brown mite, oriental mite, oriental red mite, oriental spider mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

Reason for Inclusion in Manual

Pest Description
Eggs: The eggs of *E. orientalis* are oval or circular (Fig. 1) and flattened, coming to a point dorsally but lacking the long dorsal stalk of other spider mites. Newly laid, the eggs are bright and hyaline, but later they take on a yellow, parchment-like color (Smith-Meyer, 1981).

Figure 1. Eggs (left) and adult (right) *E. orientalis*. Photos courtesy of Pedro Torrent Chocarro.

Larvae: Average size of the larva of *E. orientalis* is 190 x 120 µm. The protonymph is pale-brown to light-green, with legs shorter than the body, average size 240 x 140 µm. The deutonymph is pale-brown to light-green, average size 300 x 220 µm.

Adults: Adult female *E. orientalis* are broad, oval and flattened. They vary in color from pale brown through brownish-green to dark green with darker spots within the body. The legs are about as long as the body and yellow-brown (Fig. 1, 2). Average size is 410 x 280 µm.

Adult male *E. orientalis* are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length). The body setae are short and cannot be seen with a 10x lens (Dhooria and Butani, 1984; Smith-Meyer, 1981).

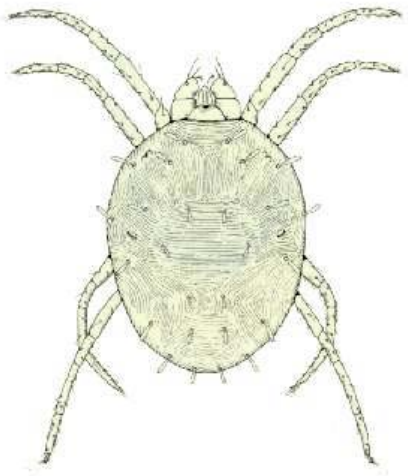


Figure 2. *Eutetranychus orientalis.* Drawing courtesy of CSIRO Entomology, Australia.

Identification requires examination of cleared and mounted female specimens by transmitted light microscopy. Diagnostic descriptions are given by Jeppson et al. (1975) and Smith-Meyer (1987).

E. orientalis has the following combination of characters: striae on the prodorsum longitudinal and tuberculated; striae between the second (d/sub/1) and third (e/sub/1) dorsocentral setae longitudinal or V-shaped; the 13 pairs of dorsal body setae all arise from basal tubercles and vary in length and shape; dorsolateral setae on the body (c2), (d2), (e2), (f2), are long, lanceolate and subspatulate or broadly spatulate; dorsocentral setae (c1), (d1), (e1), (f1), (h1) short and spatulate or lanceolate or subspatulate; first pair of dorsocentral setae (c1), first pair of dorsal lateral setae (c2) and humeral setae (c3) all more or less in line; third (e1) and fourth (f1) dorsocentral setae form a square; terminal sensillum (spinneret) of palptarsus three times as long as broad; coxa II with one seta; tactile setal formulae (I-IV): femora 8-6-(3-4)-(1-2), genua 5-5-2-2, tibiae 9-6-6-7; chromosome number (n)=3.

Pest Importance

E. orientalis is generally regarded as an important pest of citrus. In India, of the seven species reported as pests on citrus, only *E. orientalis* was reported as a major pest in all areas (Dhooria and Butani, 1984).

Symptoms/Signs

E. orientalis begins feeding on the upper side of the leaf along the midrib and then spreads to the lateral veins, causing the leaves to become chlorotic. Pale yellow streaks develop along the midrib and veins. Little webbing is produced. In heavier infestations, the mites feed and oviposit over the whole upper surface of the leaf. Very heavy infestations on citrus cause leaf fall and die-back of branches, which may result in defoliated trees. Lower populations in dry areas can produce the same effect.

Known Hosts

The primary host of *E. orientalis* is *Citrus* spp. Other hosts include *Prunus persica* (peaches), *Pyrus* spp. (pears) , *Plumeria* spp., *Cydonia oblonga* (quinces), *Ricinus communis*, *Glycine max* (soybeans), *Helianthus annuus* (sunflowers), *Ipomoea batatas* (sweet potatoes), *Eichhornia crassipes* (water hyacinth), *Citrullus lanatus* (watermelons), and over 50 other plant species. In China, *E. orientalis* attacks *Alstonia glaucescens*.

Major hosts

Abelmoschus esculentus (okra), *Carica papaya* (papaw), *Citrus* spp., *Codiaeum variegatum* (croton), *Ficus carica* (common fig), *Gossypium* spp.(cotton), *Morus alba* (mora), *Nephelium lappaceum* (rambutan), *Plumeria* (frangipani), *Ricinus communis* (castor bean), *Solanum melongena* (eggplant)

Minor hosts

Manihot esculenta (cassava), *Musa x paradisiaca* (plantain), *Olea europaea subsp. europaea* (olive), *Prunus dulcis* (almond), *Psidium guajava* (common guava)

Known Distribution

Asia: Afghanistan, Bangladesh, China, India, Iran, Israel, Japan, Jordan, Lebanon, Pakistan, Philippines, Thailand, Turkey, Yemen, **Europe:** Cyprus, Spain, **Africa:** Cape Verde, Egypt, Kenya, Malawi, Mauritania, Mozambique, Nigeria, Senegal, South Africa, Sudan, Swaziland, and **Oceania:** Australia (CABI, 2004).

Potential Distribution Within the US

No information available at this time.

Survey

The presence of *E. orientalis* can be detected by discoloration of the host leaves and pale-yellow streaks along the midribs and veins. Adult females are larger than the males. They are oval and flattened and are often pale brown through brownish-green to dark green

Key Diagnostics

Information is not available at this time.

Tetranychus kanzawai

Scientific Name
Tetranychus kanzawai Kishida

Synonyms:
Tetranychus hydrangeae

Common Name(s)
Kanzawa spider mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnica, **Order:** Acarina, **Family:** Tetranychidae

Reason for Inclusion in Manual

Pest Description
Adult (Fig. 1) females are carmine. Body 519 µm long, 313 wide. Terminal sensillum on palptarus less than 1.5 times as long as wide. Lobes of dorsal striae are taller than wide. The tibia of leg I has 9 tactile setae and that of leg II has 7. For identification and detailed description, see Wang (1981) and Tseng (1990).

Figure 1. Adult T. *kanzawai.* Photo courtesy of 1703Hhttp://ps85.nises.affrc.go.jp/~hinomoto/mites/tetranychidae.html

Pest Importance
T. kanzawai is a major pest of tea, eggplant (*Solanum melongena*) and other crops in Japan and southern China. Wang (1981) reported that it caused serious damage to cherries in China. This species is an important pest of eggplant in Taiwan (Ho and Chen, 1992). It was recently recorded on cassava in Congo (Gutierrez and Bonato, 1994) and in the Philippines (Villacarlos and Vasquez, 1988).

The cost of chemical control of *T. kanzawai* on strawberries in Taiwan exceeds US $233/hectare (Chang and Huang, 1995).

Symptoms/Signs

T. kanzawai mites and webbing are often found on the under surface of the leaves. Damaged leaves have yellowish spots. Heavily infested leaves may become yellow and dry.

Known Host

Arachis hypogaea (peanut), *Camellia sinensis* (tea), *Citrus* spp., *Fragaria ananassa* (strawberry), *Glycine max* (soybean), *Humulus lupulus* (hop), *Malus domestica* (apple), *Morus alba* (mora), *Prunus avium* (sweet cherry), *Prunus persica* (peach), *Pyrus communis* (European pear), *Solanum melongena* (eggplant), *Vitis vinifera* (grape)

Known Distribution

Tetranychus kanzawai is found in Asia, Africa, and Oceania. In North America, it is recorded in Mexico (EPPO, 2005).

Potential Distribution Within the US

No information is available at this time

Key Diagnostics

The barb of the aedeagus in *T. kanzawai* is very similar to that of *T. cinnabarinus*, but it is larger with a rounded anterior portion and acutely angled posterior portion.

No morphological differences can be found between *T. kanzawai* and *T. hydrangeae*, which were recently synonymized by Navajas et al. (2001) using ribosomal ITS2 sequences and cross-breeding experiments.

Moths

Anticarsia irrorata

Scientific Name
Anticarsia irrorata Fabricius, Walker

Synonyms:
Azazia rubricans, Thermesia rubricans

Common Name(s)
Noctuid moth, owl moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Figure 1. *A. irrorata* adult. Photo courtesy of G. McCormick., Cook Islands Natural Heritage Trust, Rarotonga.1687Hhttp://cookislands.bishopmuseum.org/species.asp?id=7003

Reason for Inclusion in Manual

Pest Description
Larvae: The larvae are yellowish green with a yellowish line on the lateral sides, a transparent mid-dorsal line and yellowish intersegmental lines. Thin and cylindrical larvae measure 4 to 4.5 cm length on full growth. After about 20 to 25 days, they undergo pupation under leaf debris. Pupal period lasts for 7 to 10 days.

Adults: Adult insect (Fig. 1) is a medium sized (15 to 17 mm) buff or light brown colored moth with an oblique transverse faint brown line across both wings dorsally. Fore-wings characterized by diagonal line from wing apex to approximately 1/3 in from outer margin; row of black dots between line and wing edge; kidney shaped cell patch approximately half way along wing. Hind wings

have continuation of diagonal line and similar subterminal dots. Underside buffish brown with subterminal line not originating in wing apex; brown dots also present and white cell spot. Well marked specimens also have wavy terminal line. Head, thorax, abdomen, legs and antennae similar shade of brown to wing background.

Pest Importance

In the field experiments conducted at National Pulses Research Centre, the defoliation of two varieties of cowpea by this moth ranged from 10.0 to 100% during December and February, 1999-2000 in Pudukkottai, Tamil Nadu, India. The corresponding yield loss was nearly 50% with a severe infestation (GPDD, 2006).

Symptoms/Signs

The larval stage of *A. irrorata* feeds on leaves. The damage can be easily recognized on foliage. The leaf margins are eaten away by the caterpillar. The caterpillar can be seen mainly on the leaf under-surface. Severely affected plants will look like a mass of veins and stems alone as if grazed by cattle.

Known Hosts

Major hosts

Andropogon sorghum (broomcorn), *Cajanus cajan* (pigeon pea), *Canavalia ensiformis* (horsebean), *Cicer arietinum* (chick pea), *Cucumis sativius* (cucumber), Cyamopsis *tetragonoloba* (cluster bean), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Lablab purpureus* (hyacinth bean), *Mucuna pruriens* (velvetbean), *Oryza sativa* (rice), *Phaseolus* spp. (bean), *Saccharum officinarum* (sugar cane), *Vigna unguiculata* (cowpea), and *Vigna* spp.

Known Distribution

Cook Islands, Fiji, French Polynesia, India, Indonesia, Malaysia (West), Nigeria, and Sri Lanka.

Potential Distribution Within the US

Information is not available at this time.

Survey

There is limited information available on this pest at this time. Survey appears to be visual based on host symptoms and presence of larvae on the leaf surface.

Key Diagnostics

Information is not available at this time.

Autographa gamma

Scientific Name
Autographa gamma Linnaeus

Synonyms:
Phytometra gamma and *Plusia gamma*

Common Name(s)
Beet worm, Silver-Y moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae,

Reason for Inclusion in Manual

Pest Description
Eggs: Semi-spherical, 0.57 mm in diameter. Eggs are yellowish-white, later turning yellowish-orange to brown. The number of ribs varies from 28 to 29 (Paulian et al., 1975).

Larvae: The larva is a semi-looper with three pairs of prolegs. It occurs in varying shades of green (Fig. 2), with a dark-green dorsal line and a paler line of whitish-green on each side. The spiracular line is yellowish, edged above with green. There are several white transverse lines between the yellow spiracular line and the dorsal black line. Some larval forms have a number of

Figure 1. Adult showing the silver *Y* mark that resembles the Greek letter gamma. Photo courtesy of Jeremy Lee.

white spots. The head has a dark patch below the ocelli or can be entirely black. Maximum length 20 to 40 mm (USDA, 1958; Jones and Jones, 1984; Emmett, 1980; Hill, 1987).

Pupae: Pupation takes place within a translucent, whitish cocoon spun amongst plant foliage. The leaves may sometimes be folded over. The pupa is brown to black, greenish or even whitish-green on its ventral side, 16 to 21 mm long, and 4.5 to 6.0 mm broad. Cremaster globular, with four pairs of hooklets (Paulian et al., 1975; Carter and Hargreaves, 1986).

Adults: The adults are grey-colored and the forewings are marbled in appearance; their color being silvery-grey to reddish-grey to black with a velvety sheen. Wing expanse is 36 to 40 mm. The 'Y' mark on the forewing is distinct and silvery (Fig. 1). The hindwings are brownish with a darker border (USDA, 1958; Jones and Jones, 1984; Hill, 1987).

Biology and Ecology

A. gamma is a migratory species and adults undertake seasonal migrations to areas where they are unable to breed continuously. In areas where it is unable to overwinter, severe infestations occur sporadically. In the United Kingdom, 1936 and 1947 were years when infestations were particularly severe and crops such as sugar beet suffered severe defoliation in July and August (Jones and Jones, 1974). More recently, there was a large migration of moths to northern Europe in 1996 (Vos and Rutten, 1998; Legrand and Wauters, 1997).

Figure 2. Larva of *A. gamma*. Photo courtesy of P. Mazzei. 1688Hwww.invasive.org

Female moths take nectar from flowers and can often be seen feeding during the day or early evening (Kwak and Velterop, 1997). They can lay from 500 to more than 1000 whitish eggs (Hill, 1987), which are laid, singly or in small batches, on the underside of leaves of low-growing plants. In temperate regions, hatching may take 10 to 12 days (Hill, 1987). The incubation period lasts for 3 days at 25°C (Ugur, 1995).

The young caterpillars feed on the foliage of their host plants and tend to occur singly, rather than in groups. When they are young, they skeletonize the leaves, but older caterpillars eat the whole leaf (Hill, 1987). Larval development takes from 51 days at 13°C to 15 to 16 days at 25°C; and, pupal development from 32 days at 13°C to 6 to 8 days at 25°C (Hill and Gatehouse, 1992; Ugur 1995). When the larvae are disturbed, they drop off the plant.

Local distribution, reproductive potential and migration are determined to a considerable extent by the availability of suitable wild plants in a given area, and good weed control reduces the threat of outbreaks. Mortality in the egg stage and the first larval instar is lowest at high humidities; mass outbreaks are known to have occurred mainly during periods of very wet weather (Maceljski and Balarin, 1974).

In areas where *A. gamma* is able to survive the winter, it overwinters as third to fourth larval instars (Tarabrina, 1970; Kaneko, 1993b; Saito, 1988) or in the pupal stage (Dochkova, 1972). There is no true diapause (Tyshchenko and Gasanov, 1983).

Pest Importance

Outbreaks of *A. gamma* occur periodically over wide areas of Europe, Asia and North Africa. Infestations were unusually heavy in flax and truck crops throughout European USSR in 1922. The outbreak of 1928, which occurred in most of central Europe, caused widespread defoliation of peas in Poland. Damage from this insect and *Pieris rapae* in areas of the Netherlands ran at as much as 320,000 guilders during some years in the 1800s. It is also very destructive in England and Denmark. Outbreaks are more frequent in North Africa and southern USSR than in central Europe. Between years of high populations, the pest is generally inconspicuous (CABI, 2004).

A. gamma occurs every year in Belgium but generally causes little damage. However, an outbreak in 1996 was particularly severe (Legrand & Wauters, 1997a,b). It is usually one of the less important caterpillar pests of *Brassica* spp. in Germany (Forster et al., 1992).

Apart from damaging the foliage of their host plants, larvae can scrape the skin from grapes and feed on the contents of the fruits. A single larva could damage 20 or more mature grapes (Abdullagatov and Abdullagatov, 1986).

Damage to globe artichokes was severe near Bari, Italy from 1982 to 1985, with about 55% plants being damaged; *A. gamma* was one of the major pests (Ippolito and Parenzan, 1985).

Studies in Czechoslovakia (Novak, 1975) indicated that damage became of economic significance when 25% of the leaf area of a plant was destroyed. Therefore, the critical density of larvae was the number of larvae/unit area required to destroy 25% of the leaf area of a plant. This varied according both to the instar of the larvae and to the development stage of the plant. The numbers of larvae/plant causing 25% leaf loss varied from 0.07 when the plant had only two leaves to 20 when it had 30 leaves.

Symptoms/Signs

Leaves may be skeletonized by larval feeding. Frass may or may not be visible.

Known Hosts

This polyphagous pest is found on cereals, grasses, fiber crops, *Brassica* spp. and other vegetables including legumes. *A. gamma* can feed on at least 224 plant species, including 100 weeds, from 51 families (Maceljski and Balarin, 1972).

Major hosts

Beta vulgaris (beetroot), *Beta vulgaris var. saccharifera* (sugarbeet), *Borago officinalis* (borage), *Brassica oleracea var. capitata* (cabbage), *Brassica oleracea* var. *gemmifera* (Brussels sprouts), *Brassica rapa* subsp. *chinensis* (Chinese cabbage), *Brassica rapa* subsp. *pekinensis* (Pe-tsai), *Cannabis sativa* (hemp), *Capsicum* (peppers), *Chrysanthemum indicum* (chrysanthemum), *Cicer arietinum* (chickpea), *Cichorium intybus* (chicory), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Glycine max* (soybean), *Gossypium* (cotton), *Helianthus annuus* (sunflower), *Hyssopus officinalis* (hyssop), *Lactuca sativa* (lettuce), *Linum usitatissimum* (flax), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Pelargonium zonale* hybrids, *Petroselinum crispum* (parsley), *Solanum tuberosum* (potato), *Spinacia oleracea* (spinach), *Trifolium pratense* (purple clover), *Triticum aestivum* (wheat), *Vitis vinifera* (grape), *Zea mays* (maize), and *Zinnia elegans* (Zinnia)

Known Distribution

A. gamma is widely distributed throughout all of Europe, and eastward through Asia to India and China; it also occurs in North Africa (USDA, 1958).

Potential Distribution Within the US

The likelihood and consequences of establishment by *A. gamma* have been evaluated in a pathway-initiated risk assessment. *Autographa gamma* was considered highly likely of becoming established in the U.S. if introduced; the consequences of its establishment for U.S. agricultural and natural ecosystems were also rated high (i.e., severe) (Lightfield, 1997). The currently reported global distribution of *A. gamma* suggests that the pest may be most closely associated with deserts and xeric shrublands, montane grasslands [not in the U.S.], and temperate broadleaf and mixed forests. Consequently, Venette et al. (2003) estimated that approximately 48% of the continental U.S. would be suitable for *A. gamma*.

Survey

Light traps have been used to monitor adult *A. gamma* (Ionescu, 1986; Zanaty et al., 1984; Kitamura et al., 1989). Much research has been done also to develop pheromone traps for monitoring *A. gamma* adults. Pheromone traps are now widely available and the use of light traps has been superseded in most cases. The pheromone lures have a high specificity (Crepin and Trouve, 1998; Kaneko et al., 1990; Kitamura et al., 1989). The optimum height for traps is 1.5 meters

above the crop (Terytze et al., 1987).

Risk forecasting based on pheromone trap catches is not effective (Crepin and Trouve, 1998). In Yugoslavia, adult flights of *A. gamma* depended to a large extent on rainfall, so that large numbers of adults could be caught, although damage caused by the larvae was below an economic level (Radin and Tosev, 1983). Inspection of plants is the most effective way of monitoring the size of infestations. Threshold levels are sometimes used (e.g. 3 to 4 larvae/plant for sugar beet). In Russia, for the silver-Y moth, soil and vegetation sampling are used to establish the extent of larval occurrence, numbers, parasitism, and to assess migratory intensity, fecundity, crop infestation and damage (Anon, 1986; CABI, 2004).

Taken from Venette et al. (2003). The USDA (1986) provides some considerations for visual inspections of host plants for the presence of eggs, larvae, or pupae. In general, eggs may be found on the lower and upper surfaces of leaves. Larvae are likely to be found, if left undisturbed, on leaves that have been skeletonized or that have holes in the interior. Pupae may be found on the lower leaf surface (USDA, 1986).

The sex pheromone, (*Z*)-7-dodecenyl acetate and (*Z*)-7-dodecenol in ratios from 100:1 to 95:5, has been used to attract and monitor male flight of *A. gamma*. In field applications, the pheromone may be dispensed from rubber septa at a loading rate of 1 mg (CAPS, 1996). Lures should be replaced every 30 days (CAPS, 1996). Newly-emerged adult males of *A. gamma* are not attracted to the pheromone; 3-day old males are most responsive to the lure. The pheromone of *A. gamma* may also attract other Lepidoptera in the U.S. such as *Anagrapha ampla*, *Anagrapha falcifera*, *Autographa ampla*, *Autographa biloba*, *Autographa californica*, *Caenurgia* spp., *Epismus argutanus*, *Geina periscelidactyla*, *Helvibotys helvialis*, *Lacinipolia lutura*, *Lacinipolia renigera*, *Ostrinia nubilalis*, *Pieris rapae*, *Polia* spp., *Pseudoplusia includens*, *Rachiplusia ou*, *Spodoptera ornithogalli*, *Syngrapha falcifera,* and *Trichoplusia ni.*

Sticky traps (e.g. Traptest traps) are relatively ineffective at capturing *A. gamma*. Modified versions of an inverted cone trap (similar to Hartstack traps) baited with 0.1 mg of (97:3) *E*:*Z*-11-tetradecenyl acetate, a general attractant of several pest species of moths, captured 30 to 135 times more *A. gamma* than did sticky traps (Burgio and Maini, 1995).

Adult males and females have also been collected using Robinson black-light traps, but these traps attract moths non-discriminately. Such traps, placed 3 meters above the ground, have been used to successfully monitor the dynamics of *A. gamma* and other Noctuid moths.

Key Diagnostics

Several life stages of three other noctuid pests can be confused with *A. gamma*. Of these, the most important species is *Trichloplusia ni*, as it is already present in the continental U.S. (Venette et al., 2003) The other easily confused species are *Cornutiplusia circumflexa* (Essex Y), which is geographically distributed in Europe, Asia and Africa and *Syngrapha interrogationis* (Scarce Silver Y), which is established in the United Kingdom. Adults of *A. gamma* are grey to grayish brown in color with a 'Y mark or gamma (γ) on the forewing' (Venette et al. 2003). Nazmi et al. (1981) compare similarities and differences between closely related species. Species are most reliably identified by close examination of the genitalia (Nazmi et al., 1980; USDA, 1986).

Chrysodeixis chalcites

Scientific Name
Chrysodeixis chalcites Esper

Synonyms:
Autographa chalcites, Chrysodeixis chalcytes, Noctua chalcites, Noctua chalcytes, Noctua chalsytis, Noctua questionis, Phalaena chalcites, Plusia buchholzi, Plusia chalcites, Plusia chalcytes, Plusia cohaerens, Phytometra chalcites

Common Name(s)
Golden twin-spot moth, green garden looper, green looper, green semi-looper. groundnut semi-looper, tomato leafworm, tomato looper

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual

Pest Description
Eggs: Eggs are white to pale green and shiny. They are dome-shaped with 28 to 32 vertical ribs from the micropyle to the base (Bretherton, 1983; Goodey, 1991).

Larvae: Mature larvae are 34 to 38 mm long, pale yellow-green with a glassy green to grey head edged with a black streak (Fig. 1). Above the spiracles on each side of the body is a thin dark green or black line stretching from the head to the seventh abdominal segment, below this is a

Figure 1. Larva of *C.chalcites*. Photo courtesy of 1691HPaolo Mazzei. 1692Hwww.invasive.org.

thicker white line from the head to the tip of the anal proleg. Spiracles are black. The ventral region is speckled with white dots (Haggett, 1980; Bretherton, 1983; Passoa, 1995; Porter, 1997). Larvae have only three pairs of prolegs, instead of the normal five, resulting in the looping gait giving rise to some of the common names. Haggett (1980) provides a detailed description and color illustration of the final larval instar.

Pupae: The pupa is 20 mm long, black in a white cocoon (Fig. 2), which turns brown then black (Harakly and Farag, 1975; Bretherton, 1983; Sannino et al. 1988).

Adults: The adult wingspan is approximately 40 mm. The forewing is 15 to 17 mm, usually gold, although some individuals have more of a bronze color (Fig. 3). There are two oval silver spots on the forewing, although in some individuals these are united. The hindwing is more pale. There are two prominent crests on the thorax (Pinhey, 1979; Bretherton, 1983; Passoa, 1995).

Figure 2. Pupae of *C. chalcites*. Photos courtesy of (left) Ernst Neering (CABI, 2004) and (right) 1689HPaolo Mazzei. 1690Hwww.invasive.org.

Biology and Ecology

C. chalcites is a polyvoltine species, with up to eight or nine generations per year in Egypt (Rashid et al., 1971). After emergence, females mate then begin oviposition within 2 or 3 days (Gasim and Younis, 1989). Eggs are laid on upper and lower leaf surfaces at night, whilst females are on the wing. Females only briefly touch the leaf to deposit one, two or a few eggs at a time. Eggs are very widely scattered in the crop (Linden, 1996). At 20°C egg incubation lasts between 5 and 26 days (Gaumont and Moreau, 1961).

Reports in the literature show considerable variation in the number of eggs oviposited by *C. chalcites.* Harakly and Farag (1975) reported females lay from 14 to 281 eggs with a mean of 149. In contrast, Gasim and Younis (1989) reported the mean number of eggs laid per female to be much higher with 385, 640 and 405 eggs at 20, 25 and 30°C, respectively.

Figure 3. Adult *C. chalcites*. Photo courtesy of 1693HPaolo Mazzei 1694Hwww.invasive.org.

Gasim and Younis (1989) studied the development rate of *C. chalcites* eggs at three temperatures, 20, 25 and 30°C. The mean length of time between oviposition and egg hatch decreased with increasing temperature. At the lower temperature eggs took 4.5 days to hatch, at 25°C they took an average of 3.0 days and at the upper temperature they took 2.0 days.

First-instar larvae graze on the underside of leaves feeding on parenchyma. They can be quite difficult to detect. A larva will drop from the leaf and hang on a silken thread if disturbed (Goodey, 1991). During the second and third instars, the larva begins to roll the edges of the leaves together and silken threads are spun on infested leaves (Rashid et al., 1971). Later instars eat through the leaves making infested leaves appear skeletonized. The last two larval instars are the most voracious feeders and will usually eat the entire leaf but may avoid the midrib, or other large veins. On legumes, they may excavate deep into pods, sometimes cutting them in two. At the optimal temperature of 25°C, there are six larval instars, each lasts approximately 2.5 to 3.5 days (Rashid et al., 1971; Harakly and Farag, 1975).

The mature larva stops feeding and enters a prepupal stage. It spins a cocoon within which it pupates. The cocoon is usually attached to the underside of a leaf but can be in the soil (Harakly and Farag, 1975). Gaumont and Moreau (1961) reported that the pupal period lasted 15 to 26 days, although at the optimal temperature of 25°C it averages 8.8 days (Rashid et al., 1971).

Adults emerge and soon begin to fly and mate. They rest with their wings folded over their back like a tent. Adults are semi-nocturnal and usually avoid strong sunlight. Generations continually breed through the year with no diapause. There are nine generations per year in Egypt (Harakly and Farag, 1975).

Pest Importance

C. chalcites is a polyphagous polyvoltine species that feeds on the foliage and fruit of vegetable, fruit and ornamental crops. It is considered one of the most serious Lepidopteran pests in many countries, although quantitative data measuring damage is lacking (CABI, 2004).

C. chalcites is the major pest of tomato in Israel during the growing season (Broza and Sneh, 1994) causing considerable damage to the leaves and vegetative parts of the plant, although it does not bore into the fruit (Harakly and Farag, 1975). In Israel, it is also one of the most important noctuid pests of fodder crops, such as alfalfa and clover (Avidov and Harpaz, 1969). It also feeds on alfalfa, maize and soybean in Spain (Amate et al., 1998). In northern Italy, *C. chalcites* is one of the principal arthropod pests on soybean (Zandigiacomo, 1990); it also attacks fields of artichokes (Ippolito and Parenzan, 1985). In Egypt, *C. chalcites* is considered the most serious of all semi-looper pests attacking field fruit and vegetables. It is a serious pest of potato in Mauritius (Anon., 1984).

In protected cultivation, *C. chalcites* can occur at any time of the year and can reach high levels of infestation on vegetables and ornamental plants. It is reported as a serious pest in Bulgaria and Turkey (Loginova, 1992; Uygun and Ozgur, 1980) affecting tomato, cucumber and peppers. *C. chalcites* is one of the four main noctuid pests of greenhouse crops in Sicily (Inserra and Calabretta, 1985) and a continual pest in greenhouses in the Netherlands (Vos and Rutten, 1995) and Belgium (Veire, 1983).

Symptoms/Signs

Leaves may be skeletonized by larval feeding. Frass may or may not be visible. The last two larval instars are the most voracious feeders and will usually eat the entire leaf but may avoid the midrib, or other large veins. On legumes, they may excavate deep into pods, sometimes cutting them in two.

Known Hosts

C. chalcites is highly polyphagous, feeding on many fruit, vegetable and ornamental crops and weeds in many plant families including Acanthaceae, Asteraceae, Bignoniaceae, Boraginaceae, Brassicaceae, Convolvulaceae, Crassulaceae, Lamiaceae, Fabaceae, Malvaceae, Orchidaceae, Rosaceae, Scrophulariaceae, Solanaceae, Verbenaceae and Violaceae. It can be a pest of crops grown outdoors and in protection, including both shade and greenhouses (CABI, 2004).

Major hosts

Glycine max (soybean), *Gossypium herbaceum* (short staple cotton), *Lycopersicon esculentum* (tomato), *Nicotiana tabacum* (tobacco), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), and *Solanum tuberosum* (potato)

Minor hosts

Anethum graveolens (dill), *Arachis hypogaea* (peanut), *Aster*, *Brassica oleracea var. botrytis* (cauliflower), *Brassica oleracea var. capitata* (cabbage), *Brassica* spp., *Capsicum annuum* (bell pepper), *Chrysanthemum indicum* (chrysanthemum), *Citrus*, *Cucumis sativus* (cucumber), *Cynara scolymus* (artichoke), *Dahlia*, *Dianthus* (carnation), *Ficus carica* (fig), *Fragaria*, *Helianthus tuberosus* (Jerusalem artichoke), *Hippeastrum hybrids* (amaryllis), *Lactuca sativa* (lettuce), *Lycopersicon pennellii*, *Medicago sativa* (alfalfa), *Musa* (banana), *Pelargonium* (pelargoniums), *Salvia officinalis* (common sage), *Stachytarpheta jamaicensis* (Jamaica vervain), *Trifolium repens* (white clover), *Triticum aestivum* (wheat), and *Zea mays* (maize)

Wild hosts

Echium vulgare (viper's-bugloss), *Marrubium* spp., *Teucrium scorodonia*, and *Urtica dioica* (stinging nettle)

Known Distribution

C. chalcites is primarily distributed between 45°N and 35°S, from southern Europe and the Mediterranean and the Middle East to southern Africa (CABI, 2004).

C. chalcites immigrants from North Africa or southern Europe, borne on strong southerly winds, are sometimes recorded in central and northern Europe (Austria, Denmark, Germany, Sweden, Switzerland and the United Kingdom (UK) in the late summer or autumn (Jor, 1973; Bretherton, 1983; Hachler et al., 1998; Palmqvist, 1998, 2002). There are about 50 records of *C. chalcites* as a migrant to the UK between 1943 and 1990 (Bretherton, 1983). Outdoor breeding populations occur in Europe as far north as northern Spain and northern Italy. No successful breeding is reported outdoors in northern Europe (CABI, 2004).

Lempke (1982) and Vos and Rutten (1995) noted that *C. chalcites* is present all year round in greenhouses in the Netherlands. Veire (1993) reported populations established in greenhouses in Belgium. However, there is no evidence that *C. chalcites* can overwinter outdoors in the Netherlands (Lempke, 1982) or elsewhere in northern Europe.

Potential Distribution Within the US

Recently, a specimen of *C. chalcites* was found on *Pelargonium* (geraniums) in a greenhouse in Ohio. This pest is not known to be established in the United States (CABI, 2004).

Survey

Leaves should be examined on upper and lower surfaces for larvae. Damage symptoms, such as skeletonized or rolled leaves with webbing may be easier to detect (CABI, 2004).

Key Diagnostics

In Africa and Europe, *C. chalcites* may be confused with *C. acuta,* although *C. acuta* is larger and has a more pointed forewing. The silver spots are also larger (Bretherton, 1983). In the U.S., immigrant *C. chalcites* appear similar to *Pseudoplusia includens*. Larvae should be reared to adulthood to confirm their identity (Passoa, 1995).

Crocidosema aporema

Scientific Name
Crocidosema aporema Walsingham

Synonyms:
Epinotia aporema, Epinotia opposita, Eucosma opposita, Eucosma aporema

Common Name(s)
Bud borer, bean shoot moth, budworm

Type of Pest
Moth

Taxonomic position:
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual:

Pest Description
Crocidosema aporema was first described by Walsingham (1914) as *Eucosma aporema* from Costa Rica. Heinrich (1931) described it as *Epinotia opposite*. Peru and Clarke (1954) considered both species synonymous, and named it *Epinotia aporema*. Powell et al. (1995) transferred it to the genus *Crocidosema.*

Morey (1972) described and illustrated the larval, pupal and adult morphology of *C. aporema* (Fig. 1).

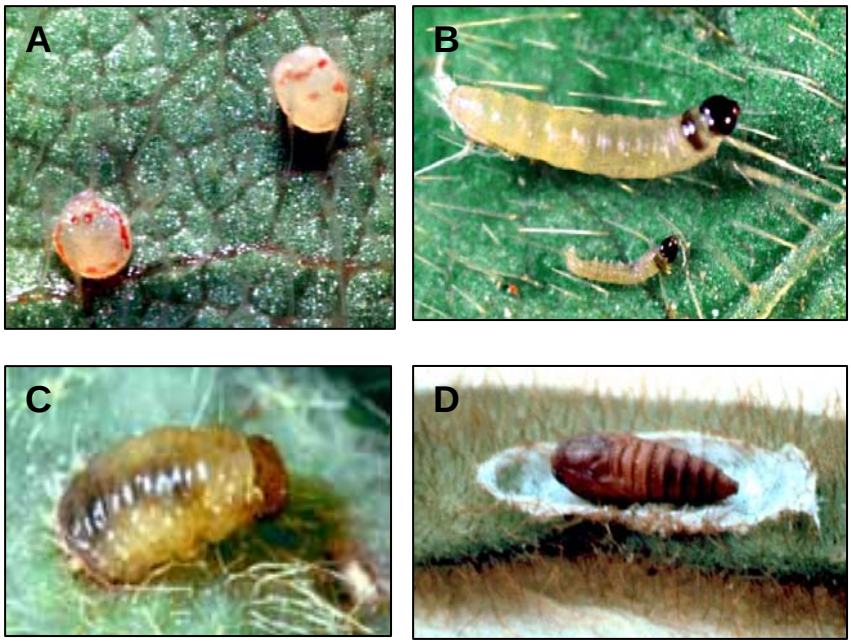


Figure 1. (A) Eggs, (B) first (smaller) and third instar larvae, (C) fifth instar larva and (D) pupa. Photos courtesy of CABI, 2004.

Eggs: Oval, 0.47 x 0.31 mm, pale yellow soon after oviposition.

Larvae: Yellowish-green, about 10 mm long when fully developed, with a shining black head capsule during the first four instars. These instars are morphologically similar, except for the size of the larva. The fully developed larva is reddish and the head capsule turns brownish in the last (fifth) instar.

Pupae Brown, 6.2 to 7.8 mm long and 1.8 to 2.2 mm wide.

Adults: Small, dark moths; forewings with a brown patterning, hindwings grey. Wingspan approximately 10 mm.

Pest Importance

C. aporema is a frequent species attacking soybeans and other Fabaceous plant species in southern Brazil, Uruguay, Chile and Argentina. Higher incidence occurs during the vegetative stages of soybeans. Plant height and insertion of the lower pods are significantly reduced as a result of its attack on terminal buds.

Symptoms/Signs

Attacked plants may be recognized by the rolled young leaflets, which contain the larvae. The larvae also tunnel along the main and secondary stems of soybean plants, drying out the terminal shoots. According to Pereyra and Sanchez (1998) the larvae may also bore into pods.

Known Host
Host records for *C. aporema* are restricted to the Fabaceae (Biezanko et al., 1974; King and Saunders, 1984).

Major hosts
Glycine max (soybean)

Minor hosts
Arachis hypogaea (peanut), *Lotus* spp.(trefoils), *Lupinus* (lupins), *Medicago sativa* (alfalfa), *Melilotus* (melilots), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), and *Vicia faba* (broad bean)

Known Distribution
C. aporema is distributed throughout the Neotropical region, including Mexico and southern U.S. Its southern range includes: Chile, Uruguay, Argentina and southern Brazil.

Potential Distribution Within the US
C. aporema is currently present in Texas.

Survey
Damage by *C. aporema* is easily detected by rolled leaflets during the vegetative stage, and by the presence of frass at the larval entrance hole in stems.

Key Diagnostics
The leaf rolling behavior of *C. aporema* can be mistaken for that of *Omiodes indicata* and *Cydia fabivora*. However, *C. aporema* attacks young leaflets, while the two other species are commonly found on fully developed leaves.

Cydia fabivora

Scientific Name
Cydia fabivora Meyrick

Synonyms:
Eulia prosecta, *Laspeyresia fabivora*, *Laspeyresia leguminis*

Common Name(s)
Torticid moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual

Pest Description
Eggs: Average size 0.89 x 0.66 mm, ventrally flattened, pale yellow initially, covered with a raised hexagonal reticulation. Red spots appear below the chorion within 24 hours of oviposition. Eventually these coalesce, and then the whole egg is red.

Larvae: Neonate larvae light orange in color; later instars have cream colored bodies and prominent prothoracic shields and heart-shaped heads. Fifth-instar larvae are approximately 18 mm long.

Pupae: Pupae have two conspicuous transverse bands of spines on abdominal sterna 3 to 9. Females are larger and heavier than males.

Adults: Adults (Fig. 1) have a 16 to 24 mm wingspan. Antenna rather stout, very shortly pubescent; head and thorax cinereous, darker on middle. Forewing roughly scaled, with several small clumps of slightly raised scales on area between base and outer third and a projecting fan of scales along inner margin near base; ground color grayish, paler at apex; markings, when distinguishable, blackish fuscous (more or less suffused in some specimens and in some

Figure 1. *C. fabivora* adult moth. Photo courtesy of Lynn Meijerman (CABI, 2004).

specimens completely so) consisting of an irregularly shaped blackish fuscous sub-tornal spot, a blackish fuscous subapical bar, divided at middle, with one arm extending towards mid termen, the other downward tornus, in some specimens the arms enclosing a contrasted, pale yellowish or orange spot, and a dark fuscous spot on outer third of cell, sometimes extending to costa and inner margin to form a dark, transverse fascia; in strongly marked specimens also an additional obscure spot, edged by slightly raised scales, just beyond cell; posterior part of costa with or without indistinct whitish strigulae; cilia pale grey, in some specimens more or less suffused with reddish ochreous. Hindwing grayish brown to brown; cilia paler. Females are generally larger than males, similar in color and markings, with more slender antennae (CABI, 2004).

Male genitalia: Valva with large cucullus, elongate triangular, densely spined toward inner (lower) margin; notch in ventral margin of valva deep. Aedeagus long, slender, curved; cornuti a cluster of short, thin, flattened spines.

Female genitalia: Anterior third of ductus bursae sclerotized, with small sclerotized collar at middle; ductus seminalis from ductus bursae just beyond the sclerotized part of tube. Corpus bursae weakly granulate, especially toward ductus bursae. Signa slender, sharp, thorn-like, with broad bases. Lamella postvaginalis consisting of a pair of elongate, triangular, sclerotized plates (CABI, 2004).

Biology and Ecology

Under laboratory conditions, adults copulate approximately 48 hours after emergence. Females start to oviposit almost immediately afterward, continuing for 2 to 4 days. Eggs are deposited singly, or occasionally in small groups, on the stems of the host plant, on the abaxial and adaxial sides of the leaves, on the petioles and pods. Generally, the preferred oviposition site shifts from leaves to reproductive structures over the course of plant development. The eggs hatch 4

to 5 days after they are deposited. Larvae have five instars (CABI, 2004).

First-instar larvae attacking plants in vegetative stages begin by perforating the stem, often at the axil of the petiole, causing desiccation of the leaf. Otherwise, the neonate larva penetrates the stem directly, leaving a short encircling mine. The larva spins a silken support and remains in the same stem until development is completed. Boring of the main stem kills small plants. Attacked pods can be identified by characteristic short brownish mines indicating where the first-instar larva has passed through to the seed. Silken support webs are also spun inside the pods, and one or two seeds are consumed, depending on seed maturity. Pupation occurs in thin cocoons, at the site of larval development in both stems and pods. The pupal stage lasts 8 to 11 days. The average time from oviposition to emergence of adults is 29.2 days (CABI, 2004).

Pest Importance

The literature on soybean pests in Brazil reports that *C. fabivora* was an important pest, although not an economically important pest. *C. fabivora* may soon be considered an economically important pest on soybean in Brazil due to a rapid increase of the pest in some areas (Foerster, 1978).

C. fabivora causes stunting of the plant and a reduction in yield. Late-maturing and late-planted varieties suffer the greatest damage from this pest. According to Stansly and Sanchez (1990), the pest could potentially build up a large population as it is able to complete three generations per crop cycle.

Symptoms/Signs

C. fabivora feeds on stems, shoots, floral buds and pods of host plants. When young plants are attacked by a larva boring into the main stem, the plant may die. Attacked pods can be identified by characteristic short brownish mines, indicating where the first-instar larva has passed through to the seed. Silken support webs are also spun inside the pods, and one or two seeds are often consumed (CABI, 2004).

C. fabivora larvae fed on soybean and *Phaseolus vulgaris* in the field. The larvae also damage the terminal shoots, passing from one shoot to another as new shoots are formed, later moving into the flower buds and causing subsequent pod loss. Severely damaged plants may become stunted, with few pods. The terminal shoots of hosts are damaged. Severely damaged plants may become stunted with few pods produced. Late-planted soybeans seem to withstand less damage than earlier planted crops (Foerster, 1978.)

Known Hosts

Major hosts

Glycine max (soybean), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean), and *Vicia fabia* (broad bean).

Known Distribution

C. fabivora is widespread throughout Central and South America including: Mexico, Costa Rica, El Salvador, Panama, Brazil, Columbia, Ecuador, Peru, and Venezuela (CABI, 2004).

Potential Distribution Within the US

Information is not available at this time. *C. fabivora* may, however, enter a country inside the stems, pods, shoots, and buds of its hosts. Specimens identified as *C. fabivora* have been intercepted from *Phaseolus spp.* three times and *Vicia faba* (seeds) once since 1975 from various Central and South American countries (USDA, 1987).

Survey

Inspect leaves, stems and pods for eggs. Cut suspect stems, pods, shoots, and buds and examine for larvae and pupae. Also inspect inside of stems for tunneling by larvae. Short encircling mines where larvae entered the stems might be visible from outside. Inspect pods for presence of larvae, pupae or webbing internally. Attacked pods may often be identified from the outside by short brownish mines where the first-instar larva passed through to the seed**.** For identification, submit suspect adult specimens, pinned and labeled to a diagnostic authority. Preserve larvae and pupae in alcohol (USDA, 1987).

Key Diagnostics

Information is not available at this time.

Etiella zinckenella

Scientific Name
Etiella zinckenella Treitschke

Synonyms:
Etiella schisticolor, Phycis zinckenella

Common Name(s)
Pea pod borer, lima bean pod borer

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family**: Pyralidae

Reason for Inclusion in Manual

Pest Description
The eggs of *E. zinckenella* are white, oval and 0.6 mm long. They are laid on young pods, the calyx or on the leaf stalk, either singly or in batches of between 2 and 12. The eggs turn pink towards the end of the incubation period of 3 to 16 days. First-instar larvae (Fig. 1) are 1 mm long with yellowish bodies and black heads. These larvae wriggle violently if their pod is opened and they are disturbed.

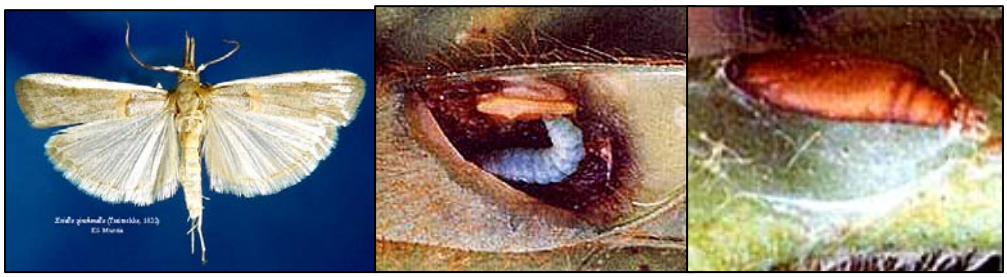


Figure 1. *Etiella zinckenella* adult male (left), larva (center), and pupa. (right). Photos courtesy of 1700Hhttp://www.nrm.se/en/svenska_fjarilar/e/etiella_zinckenella.html and 1701Uhttp://www.ayrda.org/LeC/soybean/limaborer.html

Just before pupation, larvae become green with dark-pink stripes. Full-grown larvae are 15 mm long. Freshly formed pupae (Fig. 1) are light-brown but progressively turn dark-brown to black as the time for adult emergence approaches. Male pupae are generally larger, at 8.5 mm long, than female pupae, which are 8.0 mm long. Pupae can be found in soil, 2 to 4 cm below the surface.

Adult (Fig. 1) forewings are brownish-grey with a white strip along the leading edge of the narrow forewings. The hindwings are transparent to opaque with darker outer edges. The wingspan is 24 to 27 mm.

Biology and Ecology

A single female *E. zinckenella* lays between 60 and 200 eggs during her lifetime (Kobayashi, 1976).

There are five larval instars. First-instar larvae are 1 mm long with a yellowish body and a black head. These larvae move about on the pod for half an hour; they then spin a small web, bore through the pod pericarp, which is covered by the web, and begin feeding on the developing seeds.

A number of larvae may enter the pod, but cannibalism reduces this number to only one or two. If the food supply in a pod is inadequate, the larvae migrate to another. Larval development lasts 20 days.

Full-grown larvae are 15 mm long (Kobayashi, 1976) when they leave the pod to pupate in a cocoon in the soil, 2 to 4 cm below the surface. The pupal stage lasts for between 1 and 9 weeks, depending on the temperature. After emergence, the moths live up to 20 days.

Pest Importance

E. zinckenella is a cosmopolitan pest of worldwide distribution (Qu and Kogan, 1984). Different biotypes of *E. zinckenella* exist throughout the world. For example, *E. zinckenella* is a serious pest of *Phaseolus vulgaris* in the U.S., but does not attack soybean there, despite the large area that is under cultivation. However, it is a threat to soybean in most of Southeast Asia, where it does not readily attack *P. vulgaris*.

Damage to soybean in southeast Asia is widespread. It damages about 10 to 15% of pods in Taiwan. However, up to 80% of pods may be damaged in Indonesia (Talekar, 1987). In Iloilo Province, the Philippines, where soybean is a recently introduced crop, *E. zinckenella* damaged 57% of pods, even in insecticide-protected plots (Litsinger et al., 1978).

E. zinckenella causes about 40% yield loss in soybean in the Province of Lorestan, and in adjacent areas in Iran (Parvin, 1981). In India, it infested 11.4 and 50.9% of lentil and pea pods, respectively, resulting in significant yield losses

of 10.6 and 23.9% (Singh and Dhooria, 1971). *E. zinckenella* caused 40% yield loss in cowpea in Egypt (COPR, 1981).

Symptoms/Signs

Injury to soybean pods caused by *E. zinckenella* is recognizable, even in the absence of the larvae. Large pods are marked with a brown spot where the larva has entered. As the larva develops within the pod, feces accumulate causing soft, rotten patches on the pod. Seeds are either partially or entirely eaten, and considerable frass and silk are present. A large hole is evident at the point where the larva escaped to pupate in the soil.

Blossom drop, and also some pod drop, occurs in cowpea, lentil and pigeon pea as a result of very small larvae feeding on the blossom and young pods. Usually one or two larvae can be found in each pod.

Known Hosts

E. zinckenella attacks cultivated legumes including cowpea, garden pea (*Pisum sativum*), lima bean, mung bean, pigeon pea, common bean (*Phaseolus vulgaris*) and soybean. Soybean is the preferred host.

Major hosts

Fabaceae (legumes) and *Glycine max* (soybean)

Minor hosts

Arachis hypogaea (peanut), *Cajanus cajan* (pigeon pea), *Cicer arietinum* (chickpea), *Crotalaria juncea* (sunn hemp), *Lablab purpureus* (hyacinth bean), *Lathyrus sativus* (grasspea), *Lens culinaris* ssp. *culinaris* (lentil), *Lupinus angustifolius* (lupine), *Lupinus luteus* (yellow lupine, *Medicago sativa* (alfalfa), *Pachyrhizus erosus* (yam bean), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean), *Trifolium alexandrinum* (Berseem clover), *Trigonella foenum-graecum* (fenugreek), *Vicia faba* (broad bean), *Vicia villosa*, *Vigna mungo* (black gram), *Vigna radiata* (mung bean), and *Vigna unguiculata* (cowpea)

Wild hosts

Caragana arborescens (Siberian pea-tree), *Colutea*, *Lathyrus* (Vetchling), *Phaseolus* (beans), *Robinia* (locust), *Robinia pseudoacacia* (black locust), *Spartium junceum* (Spanish broom), and *Vicia* spp. (vetch)

Known Distribution

E. zinckenella is a cosmopolitan pest of worldwide distribution (Qu and Kogan, 1984). The pest is present in Asia, Europe, Africa, North America, Central America, South America, and Australia.

Potential Distribution Within the US

The biotype of the pest that attacks bean is present in Arizona, California, Colorado, Florida, Idaho, Nevada, New Hampshire, New Mexico, Oklahoma, Rhode Island, Texas, Utah, and Washington. The biotype infecting soybean is not known to occur in the U.S. at this time.

Survey

Eggs of the insect are confined to pods and adjacent inflorescences. The minute first-instar larvae are present outside the pod for a very brief period. The presence of a fluffy locule on pods indicates that larvae are continuing to feed inside the pod. A hole in the pod pericarp indicates that the larva has already fed and has descended to the soil for pupation.

Both male and female adults are attracted to light traps. A pheromone blend consisting of tetradecyl acetate, (Z)-9-tetradecenyl acetate, (E)-11-tetradecenyl acetate and (Z)-11-tetradecenyl acetate attracts male adults of the European and Egyptian strains of *E. zinckenella* (Toth et al., 1989). The pheromone blend, which attracts the European strain of *E. zinckenella* in Hungary, is ineffective against the Southeast Asian strain of the pest.

Key Diagnostics

E. hobsoni is morphologically similar to *E. zinckenella* and also infests soybean in Indonesia. The nature of the damage caused by both species is almost identical. Naito et al. (1986) gives details of the distribution of both species in Indonesia. The morphological differences in eggs, larvae, pupae and adults are very small and are described by Naito et al. (1986). The only substantial difference that can be used to distinguish between these species is found in the adults.

The ground color of the forewing of *E. hobsoni* is dark-brown or dark-reddish brown, without the white, costal streak found in *E. zinckenella*. The antemedial transverse fascia in *E. hobsoni* is orange-edged with metallic scale. In contrast, the forewing of *E. zinckenella* varies in color from reddish-brown to purplish-grey, but is not dark, and has a white, costal streak. The antemedian transverse fascia is orange-brown to orange-red, frequently with gold iridescence.

When the adults fold their wings at rest, the antemedial bands of the forewings of *E. hobsoni* can be seen as a straight transverse band across the wings, while those of *E. zinckenella* are not straight. *E. hobsoni* is generally smaller than *E. zinckenella*; the length of the forewing of the former is 7.5 ± 0.6 mm, and that of the latter 8.7 ± 0.7 mm.

E. zinckenella may also be confused with *E. behrii*, and the two species cannot be readily separated on external characters (Whalley, 1973). In male *E. behrii*, the process on the base of the antennal segment is larger than in *E*. *zinckenella*, but both species can be easily separated on genitalic characters. *E. behrii* is widespread in Australia and probably occurs in most of Indonesia. Specimens

from peninsular Malaysia and Taiwan are slightly darker than Australian ones, but the genitalia are similar. As with all specimens in the genus, the darker color tends to fade with the increasing age of the specimen. At present, the species is known from the mainland of China, only from a few specimens in Hong Kong, although it is probably more widespread. There is less variation in the size of specimens of *E*. *behrii* than in *E*. *zinckenella*. For further information, see Whalley (1973).

Helicoverpa armigera

Scientific Name
Helicoverpa armigera Hübner

Common Name(s)
Old world bollworm, scarce bordered straw worm, corn earworm, African cotton bollworm, American bollworm, tomato worm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual

Pest Description
Eggs: Yellowish-white and glistening at first (Fig. 1A), changing to dark-brown before hatching; pomegranate-shaped, 0.4 to 0.6 mm in diameter; the apical area surrounding the micropyle is smooth, the rest of the surface sculptured in the form of approximately 24 longitudinal ribs, alternate ones being slightly shorter, with numerous finer transverse ridges between them; laid on plants which are flowering, or are about to produce flowers.

Larvae: The first and second instars are generally yellowish-white to reddish-brown in color, without prominent markings; head, prothoracic shield, supra-anal shield and prothoracic legs are very dark-brown to black, as are also the spiracles and tuberculate bases to the setae, which give the larvae a spotted appearance (Fig. 1B); prolegs are present on the third to sixth, and tenth abdominal segments. A characteristic pattern develops in subsequent instars. Fully grown larvae are approximately 30 to 40 mm long; the head is brown and mottled; the prothoracic and supra-anal plates and legs are pale-brown, only claws and spiracles remaining black; the skin surface consists of close-set, minute tubercles. Crochets on the prolegs are arranged in an arc. The final body segment is elongated.

Color pattern: a narrow, dark, median dorsal band; on each side, first a broad pale band, then a broad dark band; on the lateral line, a broad, very light band on which the row of spiracles shows up clearly. The underside is uniformly rather pale. On the basic dorsal pattern, numerous very narrow, somewhat wavy or wrinkled longitudinal stripes are superimposed. Color is extremely variable and the pattern described may be formed from shades of green, straw-yellow, and pinkish- to reddish-brown or even black.

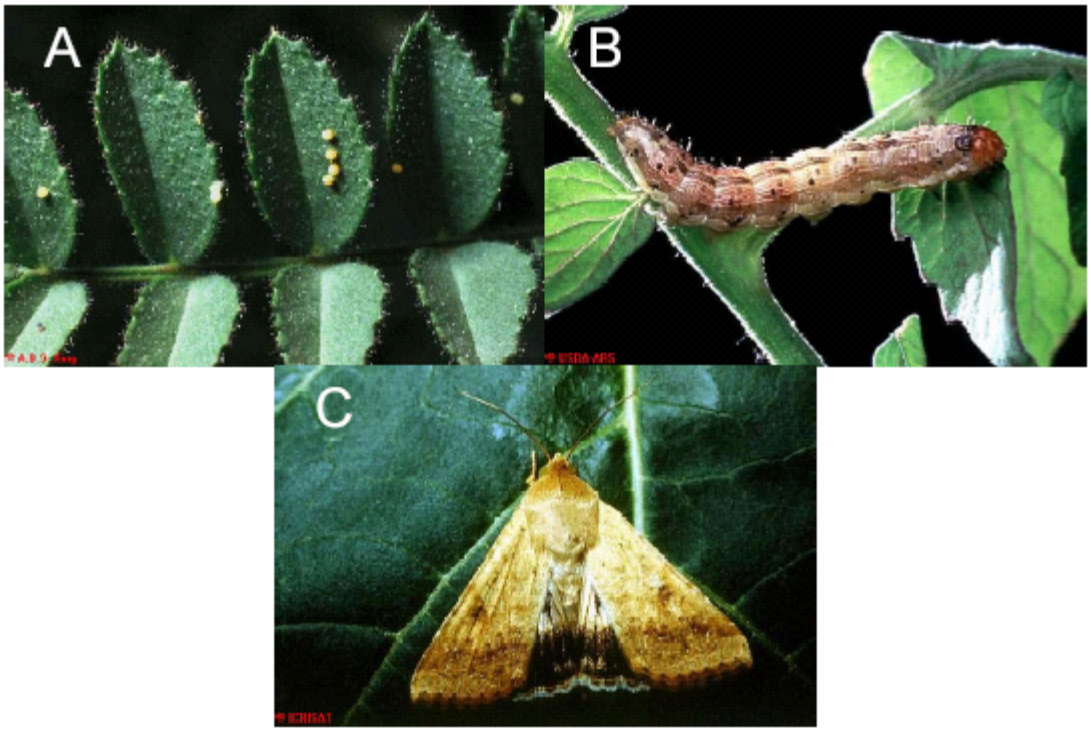


Figure 1. Life stages of *Helicoverpa armigera*, images not to scale: (A) eggs; (B) larva; and (C) adult. Photos courtesy of CABI, 2004.

Pupae: Mahogany-brown, 14 to 18 mm long, with smooth surface, rounded both anteriorly and posteriorly, with two tapering parallel spines at posterior tip.

Adults: Stout-bodied moth of typical noctuid appearance (Fig. 1C), with 3.5 to 4 cm wing span; broad across the thorax and then tapering, 14 to 18 mm long; color variable, but male usually greenish-grey and female orange-brown. Forewings have a line of seven to eight blackish spots on the margin and a broad, irregular, transverse brown band. Hindwings are pale-straw color with a broad dark-brown border that contains a paler patch; they have yellowish margins and strongly marked veins and a dark, comma-shaped marking in the middle. Antennae are covered with fine hairs.

For more information on descriptions see Dominguez Garcia-Tejero (1957), Hardwick (1965), Cayrol (1972), Delatte (1973), King (1994).

Pest Importance

H. armigera, like its close relatives *H. zea* and *Heliothis virescens* in the New World, is a pest of major importance in most areas where it occurs, damaging a wide variety of food, fiber, oilseed, fodder and horticultural crops. Its considerable pest significance is based on the peculiarities of its biology, mobility, polyphagy, rapid and high reproductive rate, and diapause. These characteristics make *H. armigera* particularly well adapted to exploit transient habitats such as man-made ecosystems. Its predilection for, and ensuing damage to, the harvestable flowering parts of high-value crops including cotton, tomato, sweet corn and the pulses confers a high economic cost. In subsistence agriculture, this results in a high socio-economic cost. However, regional and even relatively local differences in host preference can give rise to differences in pest status on particular crops. This was shown by populations in northern and southern India where severe infestations of cotton are only a relatively recent event (CABI, 2004).

Symptoms/Signs

Cotton: Bore holes are visible at the base of flower buds, the latter being hollowed out. Bracteoles are spread out and curled downwards. Leaves and shoots may also be consumed by larvae. Larger larvae bore into maturing green bolls; young bolls fall after larval damage. Adults lay fewer eggs on smooth-leaved varieties.

Tomatoes: Young fruits are invaded and fall; larger larvae may bore into older fruits. Secondary infections by other organisms lead to rotting.

Maize: Eggs are laid on the silks, larvae invade the cobs and developing grain is consumed. Secondary bacterial infections are common.

Sorghum: Larvae feed on the developing grain, hiding inside the head during the daytime. Compact-headed varieties are preferred.

Chickpea: Foliage and sometimes entire small plants consumed; larger larvae bore into pods and consume developing seed. Resistant cultivars exist.

Pigeon pea: Flower buds and flowers are bored by small larvae and may drop; larger larvae bore into locules of pods and consume developing seed. Short duration and determinate varieties are subject to greater damage. Less-preferred varieties exist.

Peanut: Leaves, sometimes flowers attacked by larvae; severe infestations cause defoliation. Less preferred varieties exist (CABI, 2004).

Known Hosts

Major hosts

Abelmoschus esculentus (okra), *Allium* (onions, garlic, leek, etc.), *Arachis hypogaea* (peanut), *Avena sativa* (oats), Brassicaeae (cruciferous crops), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Cicer arietinum* (chickpea), *Citrus*, *Cucurbitaceae* (cucurbits), *Glycine max* (soybean), *Gossypium* (cotton), *Helianthus annuus* (sunflower), *Hordeum vulgare* (barley), *Lablab purpureus* (hyacinth bean), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Mangifera indica* (mango), *Nicotiana tabacum* (tobacco), *Pennisetum glaucum* (pearl millet), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Pinus* (pines), *Pisum sativum* (pea), *Polyphagous* (polyphagous), *Prunus* (stone fruit), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (common sorghum), *Triticum* (wheat), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), and *Zea mays* (maize) (CABI, 2004).

Wild hosts

Acalypha (Copperleaf), *Amaranthus* spp. (grain amaranth), *Datura* spp., *Datura metel* (Hindu datura), *Gomphrena*, and *Hyoscyamus niger* (black henbane) (CABI, 2004).

Known Distribution

H. armigera is found in the Palearctic, Oriental, Ethiopian, and Austalian zoogeographic provinces, south of a line at approximately 52°N. The range occupied by the species includes tropical, dry, and temperate climates (CABI, 2004). The currently reported global distribution of *H. armigera* suggests that the pest may be most closely associated with deserts and xeric shrublands; Mediterranean scrub; temperate broadleaf and mixed forests; tropical and subtropical grasslands, savannas, and shrublands; and tropical and subtropical moist broadleaf forest (Venette et al., 2003).

Potential Distribution Within the US

Based on the distribution of climate zones in the U.S., approximately 49% of the continental U.S. would be suitable for *H. armigera* (Venette et al., 2003). Despite the number of *H. armigera* that are introduced into the U.S. each year, no occurrences of the pest have been reported in the wild. A wide variety of factors may contribute to the failed establishment of any introduced population, thus it is generally recognized that biological invasion is a difficult, unlikely event. It is also possible that *H. armigera* has in fact already established (conceivably small, non-damaging) populations that have gone unnoticed or been misidentified as another *Helicoverpa*/*Heliothis* species (CABI, 2004).

Survey

This polyphagous moth is one of the principal pests of cotton and maize. In Mediterranean regions, it frequently attacks vegetable plants: tomato, artichoke,

legumes, cucurbits, as well as tobacco, pinks (*Dianthus* spp.) and conifers. The caterpillar tends to be aggressive; it is carnivorous and subject to cannibalism.

The feeding larvae can be seen on the surface of plants but they are often hidden within plant organs (flowers, fruits, etc.). Bore holes and heaps of frass (excrement) may be visible, but otherwise it is necessary to cut open the plant organs to detect the pest. In temperate regions, it overwinters as a pupa buried several cm in the soil. Adults appear in April to May and can be observed until October, because of the long migration period. Females lay several hundred eggs on all parts of the plant, flowers and fruits. Eggs may hatch in less than 3 days at an optimum temperature of 27 to 28 degrees Celsius.

Visual inspections of plants for eggs and/or larvae are frequently used to monitor and assess population sizes for *H. armigera*. In vegetative Australian cotton, a minimum of 60 whole plants per 100 hectare commercial field are examined for the presence of *H. armigera* eggs or larvae; when plants begin to produce squares, only the upper terminal (approximately 20 cm) of a plant is inspected (Brown, 1984; Dillon and Fitt, 1995). In experimental plots, visual inspections for *H. armigera* in pigeon pea were restricted to the upper third of whole plants (4 sets of five plants in a 30 x 30 meter plot) (Sigsgaard and Ersbøll, 1999). Leaves of tomato plants are more attractive than flowers or fruits as *H. armigera* oviposition sites, but use of a single-leaf sample unit (with a sample size of 30 plants per field) has proven ineffective in detecting low densities of *H. armigera* (Cameron et al., 2001). On some tomato cultivars, leaves in the upper half of the plant are preferentially selected for oviposition (Saour and Causse, 1993). Larvae that are feeding on the surface of plant are easily detected, but only entry holes or frass may be visible when larvae penetrate a plant; in this case, plant dissections are needed to confirm the presence of the pest (CABI, 2004).

(Venette et al., 2003). Pheromone traps using (*Z*)-11-hexadecenal and (*Z*)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera* (Pawar et al., 1988; Loganathan and Uthamasamy, 1998; Loganathan et al., 1999; Visalakshmi et al., 2000; Zhou et al., 2000). Of three pheromone doses tested in the field (0.75, 1.0, and 1.25 mg/septum), 1 mg attracted the most males (Loganathan and Uthamasamy, 1998); the trap type was not specified. Rubber septa impregnated with these sex pheromone components (1 mg/septum) were equally effective in capturing males for 11 days in the laboratory (Loganathan et al., 1999). Captures of *H. armigera* in the field were significantly lower with 15-day-old lures than with fresh lures, and the authors recommend replacing lures every 13 days (Loganathan et al., 1999). Similar observations were reported by Pawar et al. (1988).

Trap design has a significant impact on the number of male *H. armigera* moths that will be captured with pheromone lures. Funnel traps and Texas traps are substantially more effective than sticky traps (Kant et al., 1999). Hartstack (i.e., hollow cone) traps have also been used to effectively monitor densities of adults

(Walker and Cameron, 1990). Cone traps are significantly more effective than water-pan traps (Sheng et al., 2002). Traps should be placed approximately 6 feet (1.8 meter) above the ground (Kant et al., 1999; Zhou et al., 2000), and they should be separated by a distance of at least 160 feet (50 meters) (Kant et al., 1999). For routine monitoring of pests, pheromone traps are deployed at a density of 5 traps/hectare (Sidde Gowda et al., 2002).

Adults of both sexes can be captured in black light traps.

Key Diagnostics

Several noctuid pests can be confused easily with *H. armigera*, including *H. assulta* (not known in the U.S.)*, H. punctigera (*not known in the U.S.)*, H. zea* (present in the U.S.)*,* and *Heliothis virescens* (present in the U.S.) (Kirkpatrick, 1961; CABI, 2004). Adults may be identified by distinct differences in genitalia (Kirkpatrick, 1961; Hardwick, 1965). A morphological study of *H. assulta, H. punctigera,* and *Heliothis virescens* (formerly *H. rubrescens*) compares similarities and differences between species; a key is provided for identifying adults (Kirkpatrick, 1961). Immunological tests are available to differentiate *H. punctigera* and *Heliothis virescens* in egg or larval stages (Ng et al., 1998).

The LepTon test, an Enzyme Linked Immunosorbent Assay (ELISA) based approach, has been developed to distinguish between *H. armigera* and *H. punctigera* in all stages (Trowell et al., 1993).

Lampides boeticus

Scientific Name
Lampides boeticus Linnaeus

Synonyms:
Cosmolyce baetica, *Cosmolyce boetica*, *Cosmolyce boeticus*, *Cupido boeticus*
Lampides baetica, *Lycaena baetica*, *Lycaena boetica*, *Lycaena boeticus,*
Lycaena leguminis, *Lycaenia baetica*, *Lycaenia boetica*, *Papilio damoetes*
Polyommatus baeticus, *Polyommatus boeticus*, *Papilio boeticus*

Common Name(s)
Pea blue butterfly, long tailed blue, bean butterfly, alfalfa blue, crotalaria blue, large tailed blue, pea-pod argus

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Lycaenidae

Reason for Inclusion in Manual

Pest Description
Eggs: Small (0.2 to 0.5 mm across), toroidal and china-white. Covered with fine reticulations and projections, which form an irregular pattern over the crown, and a fine network of regular ribs and knobs laterally.

Larvae: On hatching, the young larva (0.8 mm) is very active. Its citrine-yellow body is almost cylindrical with a shiny olive-colored disc on the first and last segments, and dark raised tubercles. The head is also olive, but with black mouthparts. There are two dorsal rows of short, curved, white hairs.

In the second instar, the 2.5 mm larva is pale olive-yellow and covered with minute dark tubercles; the spiracles are black. A rust-brown medio-dorsal band extends the entire body length, and there are oblique side stripes of the same color. It grows rapidly, reaching 8 mm before the next molt.

In the third and final instar, the larva is grub-like and up to 15 mm long when fully grown. It is dark green, yellowish-green or, more often, pearly-white with a yellowish tinge. All forms have a purple-brown medio-dorsal stripe, reddish lateral streaks, a brown head, short marginal hairs and a dense covering of minute setae on fine body tubercles. There is a large honey-gland on the seventh abdominal segment, which is highly attractive to some ant species (CABI, 2004).

Pupae: Wheat-grain like, 10.5 to 13 mm, smooth, minus anal hooks. The head is rounded, the meso-thorax swollen dorsally, sunken at the meta-thorax; the second abdominal segment is swollen. The abdomen then runs in a slight curve to the fourth segment and is then more abruptly curved, forming a conical anal point. It is initially of a pale flesh color, gradually turning to creamy-ochreous, or pale brown, with a darker dorsal line. Marked with varying amounts of brownish-black and covered with very fine tubercles.

Adults: Sexually dichromatic. Male wingspan 28 to 34 mm; upperside purplish-blue (Fig. 1) suffused with grey scales and with a distinct black marginal line; marginal fringe grayish-white. Female wingspan 25 to 42 mm; upperside dark brown with purple scales at the base and discal area.

Figure 1. Resting *L. boeticus* male in Kenya. Photo courtesy of A.R. Pittaway (CABI, 2004)

Both sexes have a single, 2.5 mm, black, white-tipped tail on each hindwing projecting from black tornal spots. The wing undersides are sandy-brown with creamy transverse bands. On the underside, the eye-like tornal spots are ringed with turquoise blue and crowned with orange. Antennae black, annulated white (CABI, 2004)

Biology and Ecology

Around Delhi, India, *L. boeticus* is abundant from February to April and again in November. It is least common during July and September. In Saudi Arabia, this species flies all year round, but is most common during the cooler months of November to April. The adults fly rapidly and erratically, often in groups, around the host plant. When not feeding from flowers, they frequently perch on vantage points looking for intruders, which are intercepted at high speed. *L. boeticus* is a noted migrant, capable of long distance movement, often involving vast numbers of individuals (CABI, 2004).

The eggs are laid singly on unopened flowers, sepals and flower-stalks. Several females often oviposit at the same site.

Upon hatching, the active larva tunnels into a flower or immature fruit and completes its development feeding in the immature flower or on seeds in the developing pod. In colder regions, this stage lasts 21 to 30 days. In all stages, it is very cannibalistic, devouring all competitors it comes across. It is rare to find more than one larva per pod or flower (CABI, 2004).

Pupation occurs amongst debris and leaves on the ground, under stones, or even in a curled-up, withered leaf on the plant. It is only lightly attached to a silk pad by a loose silken girdle. In areas of sandy soil, the larva may even burrow into the ground. Some individuals may even pupate in a flower, so that when the flower dies and falls to the ground, the pupa falls with it. The pupation period can last between 14 days and a year, even for caterpillars that pupated at the same time (CABI, 2004).

In Taiwan, the duration of egg, larval, pupal and adult stages were 4 to 6, 22 to 35, 6 to 9, and 2 to 5 days, respectively, with high populations observed from October to April.

In Egypt, larval feeding on *Vigna unguiculata* and *Lupinus albus* resulted in high fecundity, shorter pupal duration and greater adult longevity compared with individuals which fed on *Glycine max* or *Phaseolus vulgaris* (CABI, 2004).

Pest Importance

L. boeticus is usually a minor pest, but potentially serious. It is a major but local pest of cultivated legumes. If *L. boeticus* managed to establish itself in the New World, it would probably become a serious threat to crops. Strict controls on the movement of peas and beans in the pod need to be enforced. Importation of this species occurs each spring into central and western Europe via early green peas from the Mediterranean region (CABI, 2004).

It is the most injurious pest of *Cajanus cajan* in Cape Verde (followed by *Helicoverpa armigera* and *Etiella zinckenella*) (FAO, 1985). In Hawaii, it is a major pest of garden beans and, if not controlled, can cause considerable damage to crops locally. *L. boeticus* was captured in Hawaii before 1882. It is now the most common blue butterfly of these islands.

The incidence of *L. boeticus* on peas was studied in Haryana, India, in 1981. Damage to pods and locules averaged 8%. Larvae started to feed on the crop in the initial stages of pod formation. Peak damage was observed in the last week of March (Kaushik and Gulab Singh, 1983).

Symptoms/Signs

Larvae not only feed within developing flowers, but also on seeds within the seed pods of the host. Frass is deposited at one end of the pod, where it may cause decay. This often shows up externally as dark discoloration (CABI, 2004).

Known Hosts

Major hosts

Cajanus cajan (pigeon pea), *Canavalia* (jack bean), *Cicer arietinum* (chickpea), *Crotalaria juncea* (sunn hemp), *Glycine max* (soybean), *Lablab purpureus* (hyacinth bean), *Medicago sativa* (alfalfa), *Phaseolus* (beans), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean), *Pisum* (pea), *Pisum sativum* (pea), *Pisum sativum* var. *arvense* (Austrian winter pea), *Psophocarpus* spp., *Pueraria phaseoloides* (tropical kudzu), *Sesbania sesban* (sesban), *Vicia faba* (broad bean), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), and *Vigna unguiculata* (cowpea)

Minor hosts

Alhagi spp. (camelthorn)*, Colutea arborescens* (bladder senna), *Crotalaria pallida* (smooth crotalaria), *Gliricidia sepium* (mother of cocoa), *Indigofera* spp. (indigo), *Kennedia prostrata*, *Lathyrus odoratus* (sweet pea), *Lupinus angustifolius* (lupine), *Sesbania cannabina* (corkwood tree), *Spartium junceum* (Spanish broom), and *Virgilia oroboides.*

Wild hosts

Cytisus (Broom), *Cytisus scoparius* (broom), *Sesbania tomentosa* (ohai), *Sophora chrysophylla*, *Ulex europaeus* (gorse), *Vicia sativa* (common vetch), and *Vigna vexillata* (wild mung bean), and *Viminaria juncea (Australian native broom).*

Known Distribution

This pest is widespread throughout southern Europe, Africa, Oceania and southern Asia. It is not resident in the cooler temperate regions, such as central and northern Europe, but a few summer migrants penetrate as far north as the United Kingdom (UK) each year, with most being captured in August and September. The first confirmed specimens for the UK were captured in 1859. These butterflies appear to originate from Spain, Italy, Greece, North Africa or other Mediterranean countries (CABI, 2004)

In autumn 1971, immigrant adults of *L. boeticus* were found to have established a breeding population in the Nelson area of New Zealand. This lycaenid was first recorded in the Auckland area in 1965, and subsequently in 1967-68.

Potential Distribution Within the US

L. boeticus is present in Hawaii, but has not been recorded in the continental U.S.

Survey

The presence of adults flying around a crop, or the occurrence of the conspicuous eggs on flowers and pods, will indicate an infestation. Larvae can be found by opening discolored or malformed pods. The pest can be found in

open country, such as farms, gardens, town suburbs, woodland margins, desert oases, mountain meadows. In Europe, the pest is found mainly in flowery verges and rough places. This species avoids dense forest and is not found in rain forests.

Key Diagnostics

L. boeticus may be confused with *Syntarucus pirithous* (*Leptotes pirithous*) which is smaller with a mottled rather than streaked underside.

Leguminivoria glycinivorella

Scientific names
Leguminivora glycinivorella Matsumura

Synonyms:
Cydia glycinivorella, *Eucosma glycinivorella*, *Grapholita glycinivorella*, *Laspeyresia glycinivorella*

Common Name(s)
Soybean pod borer, soybean moth, soybean pod moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Figure 1. *L. glycinivorella* adult male Photo courtesy of Meijerman and Ulenberg.

Reason for Inclusion in Manual

Pest Description
The Tortricidae are among the largest families of the so-called micro-lepidoptera, with over 5000 species described worldwide. In North America, there are approximately 1,200 described species (Triplehorn and Johnson, 2005). Tortricidae members are more commonly found in temperate and tropical upland regions than in the lowland tropics (Meijerman and Ulenberg, 2000). Many members of this family are leafrollers. Moths in this family are small, and gray, tan or brown in color with dark bands or mottled wing areas. Front wings are often square-tipped. Wings are held roof-like over the body when at rest (Triplehorn and Johnson, 2005).

L. glycinivorella was first described by Matsumura in 1900. In Asia, *L. glycinivorella* is associated with late season plants and is found on pods and

seeds (Sinclair et al., 1997). Adults are small dark-colored moths (Fig. 1). *L. glycinivorella* larvae are responsible for the majority of soybean crop loss.
Eggs: *L. glycinivorella* eggs are flat and oval, measuring 0.48 x 0.35 mm. They are pearly-white when freshly laid. During development, a red spot appears, which may fuse to a pink streak (Meijerman and Ulenberg, 2000).

Larvae: The larval stage (Fig. 2) of the non-hibernating generation lasts 18 to 25 days, during which the larva undergoes five instars. When young, larvae are orange-yellow in color, changing to milky white or greenish in the third instar and turning orange or pink in the final instar. Heads are black and prothoracic shield is brownish (Meijerman and Ulenberg, 2000).

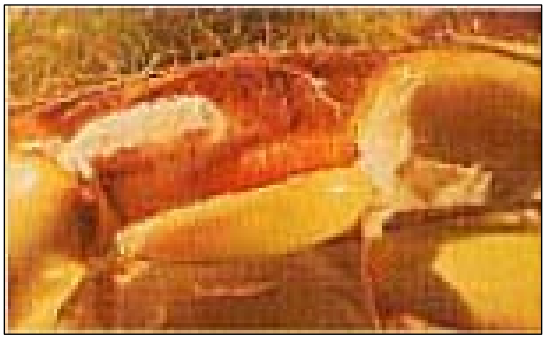


Figure 2. *L. glycinivorella* larva in East Asia. Photo courtesy of Kogan.

Pupae: Brown in color, pupae are 6 to 7mm in length (Meijerman and Ulenberg, 2000).

Adults: Adult (Fig. 3) wingspans measure 13 to 17 mm. The forewing is grey with weak purplish blue hue, becoming more yellowish near the termen. Fasciae are fuscous, irregular and narrow. Dorsal spot is well developed, giving rise to dark colored stria. Ocellus has three small black dashes. Hindwing is fuscous, and paler in color basally (Meijerman and Ulenberg, 2000).

Figure 3. *L. glycinivorella* adult in East Asia. Photo courtesy of Kogan.

Adult male (external characters): 13 to 17 mm wingspan; head and thorax ochreous-brown, abdomen fuscous. Forewing grey with weak purplish blue hue, more yellowish near termen, the latter with a slight notch. Costal strigulae brown, some giving rise to bluish striae reaching termen. Interspaces between costal strigulae yellowish. Basal, subbasal and median fasciae fuscous, irregular, narrow, angulate near costa. Interspaces between these with dark colored irregular spots. Dorsal spot well developed, fuscous, triangular, giving rise to dark colored stria; this stria connecting to stria arising from costal strigula, forming a 'T-shaped' marking. Ocellus ochreous with three small black dashes. Cilia dark yellowish. Hindwing with anal fold, fuscous, paler basally; cilia yellowish grey (Meijerman and Ulenberg, 2000).

Male genitalia: Tegumen long, broad terminally, proximal portion expanding dorsally, with long-haired patches situated laterally before apex of tegumen. Cucullus broad, somewhat expanding posteriorly; notch in ventral margin of valva rather small. Aedeagus long, curved (Meijerman and Ulenberg 2000).

Female adult (external characters): Similar forewing to male; hindwing without anal fold.

Female genitalia: Ovipositor fairly long, papillae analis small. Sterigma in form of a weakly sclerotized, indistinct lamella postvaginalis marked with some terminal hairs; ostium with short sclerite; ductus bursae long, membranous; corpus bursae with well developed signa and posterior diverticulum.

Biology and Ecology

L. glycinivorella is univoltine in northern Japan (Sakagami et al., 1985); however, a second generation has been noted in other locations. Adults emerge in late-July to early August, and females oviposit on young bean pods. Larvae, once hatched, enter pods and eat immature beans. In mid- to late-October, full grown larvae leave the pods, enter the soil and spin cocoons.

Females lay about 160 to 170 eggs each. Over 80% of the eggs are deposited on young pods. Before young pods are available, petioles and stipules are common sites. After 7 to 9 days, the eggs hatch. The larva spins a loose silken covering, probably for support when gouging out pod tissue (Meijerman and Ulenberg, 2000). A hibernating (fifth instar) larva spins a cocoon and overwinters in the soil. The larvae spend eight or more months in cocoons until pupation the following year, which occurs in July, approximately. The cocoon does not protect the larva from ultra-low temperatures, but it is thought to prevent inoculative freezing, which takes place at approximately -4.0°C. Cocoons also provide protection from submergence during early spring flooding and pre-emergence (Sakagami et al., 1985).

Pest Importance

L. glycinivorella is considered one of the most serious soybean pests in Northeast Asia (Sakagami et al., 1985).

Symptoms/Signs

Late and widely spaced planting tends to result in heavier pod-borer damage than does early and dense planting. The date of pod setting and the duration of pod ripening also appear to be related to the damage-rate (Meijerman and Ulenberg, 2000). Larvae feed on the seeds inside the pod. The entrance hole in the pod created by *L. glycinivorella* is very small, and the callus tissue formed over it resembles the feeding punctures made by pod sucking bugs. Inside the pod, the larva feeds on the seeds. The number of larvae per pod varies with pod size and host variety (Meijerman and Ulenberg, 2000).

Known Hosts

Major Host
Glycine max (soybean) and *Phaseolus* spp. (beans)

Minor Hosts:
Pueraria lobata

Wild Hosts
Lupinus spp. (lupine)

Known Distribution

L. glycinivorella is known to occur in China, Japan, Korea, and the former USSR (Meijerman and Ulenberg, 2000).

Potential distribution Within the US

Information is not available at this time.

Survey

Specific information is not available at this time. However, other moths in this family are surveyed using visual observation of symptoms, larvae, pupae, webbing, and frass.

Key Diagnostics

Small, dark colored moths. Small entrance holes on pods can be observed. Larvae may be found inside pods.

The tortricid pod-borer *Fulcrifera orientis,* collected from *Sophora flavescens* in Japan, has been confused with *L. glycinivorella* (Meijerman and Ulenberg, 2000). *F. orientis* can be differentiated based on male genitalia. The aedeagus of *F. orientis* is armed with a long process, originating from the anellus above the base of the coecum penis.

Mamestra brassicae

Scientific Name
Mamestra brassicae Linnaeus

Synonyms:
Barathra brassicae, Hypobarathra unicolor, *Noctua albidilinea, Phalaena noctua brassicae*, *Phalaena omicron*

Common Name(s)
Cabbage moth, cabbage armyworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual

Pest Description
Eggs: The eggs are relatively small, hemispherical, ribbed and reticulate. They are whitish in color when newly laid, but turn gradually to purplish-brown with a brown to purple micropyle and basal ring. A few hours before hatching, they darken to grayish-black. The eggs are laid singly in regular batches of up to 70 to 80 eggs, mainly on the undersides of leaves.

Larvae: There are six instars. First- and second-instar larvae are about 3 to 10 mm long, greenish and more or less translucent with black hairs on black warts. First-instar larvae have a black head capsule, but after the first molting it turns light-brown (Fig. 1). The prolegs on the third and fourth abdominal segments are poorly developed in the first two or three instars. From the third instar, the larvae are pale-green with yellowish intersegmental bands. The dorsal region turns gradually darker with each

Figure 1. *M. brassicae* larvae feeding on leaf. Photo courtesy of CABI, 2004.

molt, and in the last instar the majority of the larvae are brownish-green or blackish-green.

Heath and Emmet (1979) described full-grown larvae. The body is about 50 mm long, elongate, and with a slight dorsal hump on abdominal segment 8. The head capsule is light-brown, and the dorsal region of the body is from fairly bright-green, through brownish-green to almost black. The dorsal line is fine and black. On each side, there is one sub-dorsal line of blackish bars. The spiracular line is broad and pale-green or pale-ochreous. The spiracles are white. The ventral region is yellowish-green.

Pupae: The pupae are elongate, 17 to 22 mm long, and reddish-brown and glossy. The wing- and limb-cases are finely sculptured. The abdominal segments are darker brown and evenly tapered, and there is a finely pitted anterior band on each segment. Segment 8 is sharply excavated to a narrow conical cremaster with two short apically hooked spines. Pupation takes place within flimsy cocoons in the soil (Heath and Emmet, 1979).

Adults: The adult moths have a wingspan of 34 to 50 mm. The forewings are mottled and may appear grey-brown, brown or blackish-brown, with variable reddish-brown scaling. Sub-basal, antemedian and postmedian lines are inconspicuous and slightly paler than the background color, and have a fine dark edge. A kidney-shaped stigmata outlined in black with a whitish distal margin and a less clearly defined proximal margin, is placed near the center of each forewing. The subterminal line is very variable. When present it is whitish and irregular, with two angular projections (like a W). The hindwings are fuscous and generally paler than the forewings. They are light-grayish towards the base, and have a darker terminal shade. The fringe has a grayish central line. The eyes are hairy and the forelegs have a characteristic brown, slightly curved, apically pointed tibial spur. Like other species in Hadeninae, the eyes are hairy.

Pest Importance

In central parts of the distribution area *M. brassicae* is a serious pest, mainly on Brassica spp., beetroots and legumes, but also on other vegetable crops (Heath and Emmet, 1979; Filippov, 1982; Poitout and Bues, 1982; Hommes 1983; Øgaard, 1983; Kahrer, 1984; Injac and Krnjajic, 1989; Finch and Thomson, 1992; Van de Steene, 1994). In these areas, the greatest damage is usually caused by the larvae of the second generation, which are often more numerous than the first generation (Kahrer, 1984; Injac and Krnjajic, 1989). In the northern areas (Scandinavia and Finland), the occurrence as a serious pest is more sporadic (Skou, 1991; Johansen, 1997b).

In cabbage crops in Germany, *M. brassicae* is a main pest with regular occurrence. In field experiments, 27 to 98% of the plants in different cabbage crops were infested (Hommes, 1983). According to Filippov (1982) larval infestation of cabbage in Moldavia led to harvest losses of 8 to 80%. In a study of

white cabbage in Norway, weight losses due to larval damage were 10 to 13% (Rygg and Kjos, 1975). In Belgium, insecticides are often applied to Brussels sprouts every 2 to 3 weeks to control *M. brassicae* larvae (Van de Steene, 1994).

Symptoms/Signs

Small larvae feed on the underside of the external leaves, where they make small perforations. As the larvae grow older, the feeding holes become larger. Severe infestations of small larvae may rapidly skeletonize the leaves, and can sometimes destroy small plants. Older larvae tunnel into the heart of the plants. They leave considerable amounts of feces, which favor growth of decaying bacteria and fungi. Most crop losses caused by the larvae occur as a result of boring and fouling rather from the amount of plant tissue eaten. Even slight infestations of older larvae can be damaging, particularly in crops such as heading cabbage, where the larvae destroy the marketable product (Heath and Emmet, 1979; Finch and Thomson, 1992).

In cauliflower and broccoli, the larvae also feed on the inflorescence, where they chew more or less deep holes. Small larvae live well hidden between the flower stems and may pass sorting procedures, contaminating processed products.

Soybean leaves may be completely skeletonized. The feeding may destroy young buds, leading to distorted growth. The larvae bore into the pods and feed on the seeds (Lihnell, 1940).

The larvae feed on leaves, buds and petals in ornamentals such as *Dahlia*, *Chrysanthemum* and *Rosa* spp., and they may bore into the fruits in fruiting crops, such as tomato.

Known Hosts

M. brassicae larvae are extremely polyphagous: although they prefer Brassica crops (Heath and Emmet, 1979; Skou, 1991; Finch and Thomson, 1992). Beetroots, legumes, lettuces, onions and potatoes are also frequently reported to be infested (Øgaard, 1983; Injac and Krnjajic, 1989; Finch and Thomson, 1992; Zhang, 1994). The species is also found on a wide range of other vegetable crops and ornamental flowers in greenhouses and in the open, and on a wide range of deciduous tree species.

Major hosts

Allium cepa (onion), *Allium sativum* (garlic), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Brassica oleracea* (cabbages, cauliflowers), *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Brassica oleracea* var. *gemmifera* (Brussels sprouts), *Brassica rapa* subsp. *pekinensis* (Pe-tsai), *Glycine max* (soybean), *Lactuca sativa* (lettuce), *Lycopersicon esculentum* (tomato), *Nicotiana*, *Nicotiana tabacum* (tobacco), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Solanum tuberosum* (potato), and *Zea mays* (maize)

Minor hosts
Callistephus chinensis (China aster), *Capsicum* (peppers), *Capsicum annuum* (bell pepper), *Chrysanthemum* (daisy), *Dianthus caryophyllus* (carnation), *Fragaria*, *Linum usitatissimum* (flax), *Malus domestica* (apple), *Medicago sativa* (alfalfa), *Prunus persica* (peach), *Rosa* (roses), *Trifolium repens* (white clover), *Vicia faba* (broad bean), and *Vitis vinifera* (grape).

Known Distribution
M. brassicae is present throughout the Palaearctic region from Europe to Japan and subtropical Asia. According to Finch and Thomson (1992), *M. brassicae* is abundant throughout Central Europe and temperate Asia. Øgaard (1983) states that the species is present mainly between 30°N and 70°N. The species is abundant all over Denmark and in southern Scandinavia and Finland (Skou, 1991). In Norway, *M. brassicae* occurs as a pest up to 62°N (Johansen, 1997b). The species is not found on Iceland.

Potential Distribution Within the US
The species is not present in America or Oceania (APPPC, 1987; Zhang, 1994).

Survey
Adults can be detected with pheromone or light traps. Egg batches and small larvae (less than about 1.5 cm) are found mostly on the undersides of the larger external leaves. Feeding perforations from the smallest larvae are difficult to detect. Large larvae are found between the internal leaves in the heart of plants, in tunnels or cavities in cabbage heads, flowers, buds or fruits. Look for feeding holes, entrance holes and feces.

Crop scouting should be done on a number of plants per field at least weekly, and should start 1 to 2 weeks after the first adults are caught in the traps. Scouting methods have been developed and are recommended (Kahrer, 1984; Freuler, 1992; Planteforsk and ITAS, 1997).

Key Diagnostics
The adults resemble many other dull-colored members of the Noctuidae. Identification of adult noctuids is often based on characteristics of male genitalia. *Mythimna pallens, Discestra trifolii,* and *Lacanobia w-latinum* can be distinguished from *M. brassicae* by no tibial spur on foreleg. *Manilkara zapota* and *Apamea* spp. can be distinguished from *M. brassicae* by no tibial spur on foreleg, and glabrous eyes

It is difficult to distinguish between larvae from different noctuid species, especially in the youngest instars. See Heath and Emmet (1979) or Skinner (1998) for full description.

Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

Common Name(s)
Cotton leafworm, Egyptian cotton leafworm, Mediterranean climbing cutworm, tobacco caterpillar, tomato caterpillar, Egyptian cotton worm, Mediterranean brocade moth, Mediterranean climbing cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual

Pest Description
Eggs: Spherical, somewhat flattened, 0.6 mm in diameter, laid in clusters arranged in more or less regular rows in one to three layers, with hair scales derived from the tip of the abdomen of the female moth (Fig. 1). Usually whitish-yellow in color, changing to black just prior to hatching, due to the big head of the larva showing through the transparent shell (Pinhey, 1975).

Figure 1. Eggs and neonates. Eggs are laid in batches covered with orange-brown hair scales. Photo courtesy of 1702Hhttp://www.defra.gov.uk/planth/pestnote/spod.htm

Larvae: Larvae grow to 40 to 45 mm and are hairless, cylindrical, tapering towards the posterior and variable in color (blackish-grey to dark green, becoming reddish-brown or whitish-yellow) (Fig. 2). The sides of the body have dark and light longitudinal bands; dorsal side with two dark semilunar spots laterally on each segment, except for the prothorax; spots on the first and

eighth abdominal segments larger than the others, interrupting the lateral lines on the first segment. The larva of *S. littoralis* is figured by Bishari (1934) and Brown and Dewhurst (1975).

Pupae: When newly formed, pupae are green with a reddish color on the abdomen, turning dark reddish-brown after a few hours (Fig. 3). The general

Figure 2. (A) Larva of *S. littoralis*, showing light and dark longitudinal bands on sides of the body and dark spots on segments of the dorsal side (B) *S. littoralis* larva feeding on cotton leaves. Photo courtesy of CABI, 2004.

shape is cylindrical, 14 to 20 x 5 mm, tapering towards the posterior segments of the abdomen. The last segment ends in two strong straight hooks (Pinhey, 1975).

Adults: Moth with grey-brown body (Fig. 3), 15 to 20 mm long; wingspan 30 to 38 mm; forewings grey to reddish brown with paler lines along the veins (in males, bluish areas occur on the wing base and tip); the ocellus is marked by two or three oblique whitish stripes. Hindwings are grayish white, iridescent with grey margins and usually lack darker veins (EPPO, 1997).

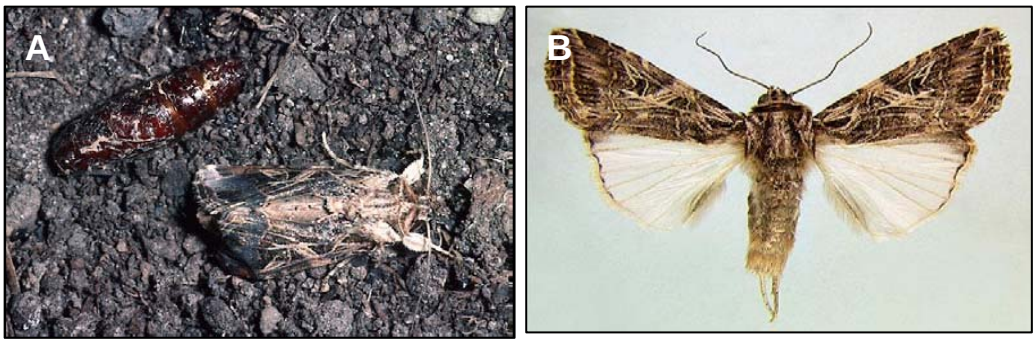


Figure 3. Pupa and adult of *S. littoralis* on soil (A). Adult moth of *S. littoralis* (museum set specimen) (B). Photos courtesy of CABI, 2004 and Entopix.

Pest Importance

S. littoralis is one of the most destructive agricultural Lepidopteran pests within its subtropical and tropical range. It can attack numerous economically important crops throughout the year (EPPO, 1997). On cotton, the pest may cause considerable damage by feeding on the leaves, fruiting points, flower buds and occasionally on bolls. When peanuts are infested, larvae first select young folded leaves for feeding, but in severe attacks, leaves of any age are stripped off. Sometimes, even the ripening kernels in the pods in the soil may be attacked. Pods of cowpeas and the seeds they contain are also often badly damaged. In tomatoes, larvae bore into the fruit and render them unsuitable for consumption. Numerous other crops are attacked, mainly on their leaves.

In Europe, damage caused by *S. littoralis* was minimal until about 1937. In 1949, there was a catastrophic population explosion in southern Spain, which affected alfalfa, potatoes and other vegetable crops. At present, this noctuid pest is of great economic importance in Cyprus, Israel, Malta, Morocco and Spain (except the north). In Italy, it is especially important on protected crops of ornamentals and vegetables (Inserra and Calabretta, 1985; Nucifora, 1985). In Greece, *S. littoralis* causes slight damage in Crete on alfalfa and clover only. In North Africa, tomato, pepper, cotton, maize and other vegetables are affected. In Egypt, it is one of the most serious cotton pests.

Symptoms/Signs

On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants. On cotton, the larvae feed on the leaves creating large holes of irregular shape and usually all that remains are the bigger veins. The larvae may also bore into the bud or young boll and consume the whole contents, causing them to be shed or dry up (Bishari, 1934). Bolls have large holes in them from which yellowish- to dark-green larval excrement protrudes. On tobacco, leaves develop irregular, brownish-red patches and the stem base may be gnawed off. Maize stems are often mined by *S. littoralis* and young grains in the ear may also be damaged.

Known Hosts

The host range of *S. littoralis* covers over 40 families, containing at least 87 species of economic importance (Salama et al., 1970). Economically important hosts include: okra, onion, beet, cabbage, cauliflower, tea, bell pepper, watermelon, *Citrus* spp., coffee, carrot, cotton, soybean, fig, sunflower, sweet potato, potato, pea, bean, rice, tomato, cereal crops, tobacco, radish, roses, sugar cane, guava, spinach, cocoa, maize, cowpea, and grape.

Known Distribution

The northerly distribution limit of *S. littoralis* in Europe corresponds to the climatic zone in which winter frosts are infrequent. It occurs throughout Africa and extends eastwards into Turkey and north into eastern Spain, southern France and northern Italy. However, this boundary is probably the extent of migrant

activity only, because although the pest overwinters in southern Spain, it does not do so in northern Italy or France. In southern Greece, pupae have been observed in the soil after November and the species overwinters in this stage in Crete. Low winter temperatures are, therefore, an important limiting factor affecting the northerly distribution, especially in a species with no known diapause (Miller, 1976; Sidibe and Lauge, 1977).

Potential Distribution Within the US

The potential U.S. range of most Spodoptera may be limited to the west coast through the lower southwestern and southeastern U.S., reaching as far north as Maryland. Migratory species may be capable of periodic spread into northern states and even Canada by late summer or early fall.

Survey

A number of sampling considerations for *S. littoralis* have been proposed: Surveys for this pest can take place any time during the growing season while plants are actively growing; early instars (<3rd) are likely to be on lower leaf surfaces during the day; larvae will skeletonize leaves by feeding on this surface and such damage to the leaf provides evidence of the presence of larvae; sweep net sampling may be effective at dawn or dusk; specimen identification should be confirmed by a trained taxonomist (Venette et al., 2003; USDA, 1982). However, not all sampling methods are equally effective for all life-stages of the insect. Eggs are only likely to be found by visual inspection of leaves. First through third instars may be detected by sweep net sampling; nearly all instars can be detected by visual inspection of plants; and, later instars (4th-6th) and pupae may be found by sieving soil samples (Abul-Nasr and Naguib, 1968; Abul-Nasr et al., 1971).

Active traps (either light- or pheromone-based) have been recommended for monitoring relative densities of adults (DEFRA, 1999). Pheromone traps can be used to monitor the incidence of *S. littoralis* (Rizk et al., 1990). The synthetic sex pheromone (*Z*,*E*)-(9,11)-tetradecadienyl acetate has proven highly effective at trapping male moths of *S. littoralis* (Salem and Salama, 1985). Kehat and Dunkelblum (1993) found that the minor sex pheromone component, (9Z,12Z)-9,12-tetradecadienyl acetate in addition to the major component (9Z,11Z)-9,11-tetradecadienyl acetate was required for to attract males. Sex-pheromone baited delta traps remained attractive for approximately 2 weeks, but effectiveness declined after 3 to 4 weeks of use (Ahmad, 1988). To monitor male flight activity in vegetable production areas, delta traps were placed 1.7 meters above the ground at a rate of 2 traps/hectare (approximately 1 trap/acre) (Ahmad, 1988). Pheromone lures impregnated with 2 mg of the pheromone blend (blend not specified) were replaced after 4 weeks of use (Ahmad, 1988).

Traps are deployed at a similar height (1.5 meters) to monitor male flight in cotton (Salem and Salama, 1985). Catches in pheromone traps did not correlate as well with densities of egg-masses in cotton fields as did catches in a black-

light trap (Rizk et al., 1990). The attractiveness of traps baited with (*Z*,*E*)-(9,11)-tetradecadienyl acetate is governed primarily by minimum air temperature; relative humidity, adult abundance, and wind velocity (Venette et al., 2003). Densities of female *S. littoralis* also affect the number of males that are captures at different times of the year (Rizk et al., 1990). Lures for *S. littoralis* can be used in the same traps with lures for *S. litura*, *Helicoverpa armigera*, *Pectinophora scutigera* (all not known to occur in the U.S.), and *P. gossypiella* (exotic established in U.S.) (Venette et al., 2003). Lures for *S. littoralis* may also attract *Erastria* spp. (established in U.S.) (PPQ, 1993).

Light traps using a 125 W mercury-vapor bulb have been used to nondiscriminately capture multiple *Spodoptera* spp. (Blair, 1974) and most assuredly other insects as well. A modified light trap using six 20-W fluorescent lights also proved an effective for monitoring flight activity of *S. littoralis* (El-Mezayyen et al., 1997).

See http://www.aphis.usda.gov/ppq/manuals/pdf_files/NPRG-Spodoptera.pdf for additional survey information.

Key Diagnostics

S. littoralis is often confused with *S. litura* and the variability and similarity of the two species makes correct identification difficult and examination of adult genitalia is often the only certain method. For more information on morphological discrimination between the adult, pupal and larval stages of the two species, refer to Schmutterer (1969), Cayrol (1972), Mochida (1973) and Brown and Dewhurst (1975). Although markings on larvae are variable, a bright-yellow stripe along the length of the dorsal surface is characteristic of *S. litura*. On dissection of the genitalia, the ductus and ostium bursae are the same length in female *S. littoralis*, whereas they are different lengths in *S. litura*. The shape of the juxta in males in both species is very characteristic, and the ornamentation of the aedeagus vesica is also diagnostic.

Spodoptera litura

Scientific Name
Spodoptera litura Fabricius

Synonyms:
Mamestra albispars, Noctua elata, Noctua histrionica, Prodenia ciligera, Prodenia declinata, Prodenia evanescens, Prodenia glaucistriga, Prodenia litura, Prodenia subterminalis, *Prodenia tasmanica, Noctua litura, Prodenia testaceoides*, *Prodenia littoralis, Spodoptera littoralis*

Common Name(s)
Armyworm, taro caterpillar, tobacco budworm, cotton leafworm, rice cutworm, cluster caterpillar, cotton worm, Egyptian cotton leafworm, tobacco caterpillar, tobacco cutworm, tobacco leaf caterpillar, common cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual

Pest Description
The two Old World cotton leafworm species, *Spodoptera litura* and *S. littoralis*, are allopatric, their ranges covering Asia and Africa, Europe and the Middle East, respectively. Many authors have regarded them as the same species, but they have been differentiated based on adult genitalia differences.

Eggs: Spherical, somewhat flattened, 0.6 mm in diameter, laid in batches and covered with hair scales from the tip of the abdomen of the female moth. Usually pale orange-brown or pink in color (Fig. 1). Egg masses measure about 4 to 7 mm in diameter and appear golden brown, because they are covered with body scales of females (CABI, 2004).

Larva: Larva hairless, variable in color (young larvae are light green, the later instars are dark green to brown on their backs, lighter underneath) (Fig. 1); sides

of body with dark and light longitudinal bands; dorsal side with two dark semilunar spots laterally on each segment, except for the prothorax; spots on the first and eighth abdominal segments larger than others, interrupting the lateral lines on the first segment. Though the markings are variable, a bright-yellow stripe along the length of the dorsal surface is characteristic of *S. litura* larvae (CABI, 2004). Larval instars can be distinguished based on head capsule width ranging from 2.7 to 25 mm, and body length ranges from 2.3 to 32 mm.

Figure 1. (Left) egg mass with parasites (center) larva, and (right) adult. Photos courtesy of CABI, 2004.

Pupa: 15 to 20 mm long, red-brown; tip of abdomen with two small spines.

Adult: Moth, with grey-brown body (Fig. 1), 15 to 20 mm long; wingspan 30 to 38 mm. The forewings are grey to reddish-brown with a strongly variegated pattern and paler lines along the veins (in males, bluish areas occur on the wing base and tip); the hindwings are grayish-white with grey margins, often with dark veins in *S. litura* (but without in *S. littoralis*) (CABI, 2004). See Schmutterer (1969), Cayrol (1972), and Brown and Dewhurst (1975) for additional information.

Pest Importance

S. litura larvae are polyphagous defoliators, seasonally common in annual and perennial agricultural systems in tropical and temperate Asia. This noctuid is often found as part of a complex of lepidopteran and non-lepidopteran foliar feeders, but may also damage tubers and roots. Hosts include field crops grown for food and fiber, plantation and forestry crops, as well as certain weed species.

S. litura is also a member of a complex that causes extensive defoliation of soybean (Bhattacharjee and Ghude, 1985). Defoliation as severe as 48.7% during the pre-bloom stage of growth caused no 'marked' difference from a control treatment in which defoliation was prevented by repeated insecticide application. Number and weight of pods and grains per plant were, however, reduced when defoliation occurred at, or after, blooming.

Most work on the economic impact of *S. litura* has been conducted in India, where it is a serious pest of a range of field crops. It has caused 12 to 23% loss to tomatoes in the monsoon season, and 9 to 24% loss in the winter (Patnaik, 1998). In a 40- to 45-day-old potato crop, damage ranged from 20 to 100% in

different parts of the field depending on moisture availability. Larvae also attacked exposed tubers when young succulent leaves were unavailable.

S. litura is also a pest of sugarbeet, with infestations commencing in March and peaking in late March and April (Chatterjee and Nayak, 1987). Severe infestations led to the skeletonization of leaves, as well as feeding holes in roots that rendered the crop 'virtually unfit for marketing'. Late harvested crops were most severely affected and, in extreme cases, 100% of the roots were damaged, leading to considerable yield reduction. Work on this species in a complex of other sugarbeet defoliators (*S. exigua* and *Spilosoma obliqua*) led to the development of an interactive exponential model based on length and severity of defoliation. It explained 88 to 90% of the variability in root and sugar yields and suggested the need for pest control when defoliation exceeded 25% during April. Control was not required if the pest appeared after the first week of May (Singh and Sethi, 1993).

S. litura is one of six defoliating pests of fodder cowpea which, in a field experiment, were responsible for consuming up to 85.5% of leaf area (Ram et al., 1989). Aroid tuber crops (including taro (*Colocasia esculenta*)) suffered yield losses of up to 29% as a result of infestation by *S. litura*, *Aphis gossypii* and spider mites (Pillai et al., 1993).

In peanut, *S. litura* is one of several pests that can be important during the pegging, podding and pod maturation stages of growth (Singh and Sachan, 1992). Several studies have aimed at quantifying the damage attributable to *S. litura*. Field experiments by Panchabhavi and Raj (1987) extended over 2 years, used artificial infestation of peanut plots of 15 m² with differing densities of *S. litura*. Infestation levels of just three egg masses (of 250 eggs each) caused significant loss of peanut pods and haulms. Infestation with 12 egg masses per plot led to a haulm yield reduction of up to 43.7% and a pod yield reduction as high as 27% compared with an insecticide-protected control treatment. In other field experiments over 3 consecutive years, leaf damage attributed to *S. litura* tended to decline with delayed sowing time irrespective of peanut cultivar (Patil et al., 1996). Leaf damage fell from 51.8% for mid-June sown crops to 19.2% for late-July sown crops. Mean pod yields were 2.68 and 0.99 tons/hectare, respectively.

S. litura causes damage to many species of forest and plantation trees and shrubs (Roychoudhury et al., 1995). It is responsible for brown flag syndrome in banana (Ranjith et al., 1997), and 5 to 10% fruit damage in grapes (Balikai et al., 1999). In Paulownia nurseries and plantations, a complex of at least 24 defoliating pest species causes damage. Within this complex, *S. litura* was considered the most important noctuid species, with an incidence of 72% in weekly surveys (Kumar and Ahmad, 1998). Peak activity occurred in July and September, with an average of 6.5 and 5.2 larvae per plant in these months, respectively. During this period, many plants were completely defoliated by *S.*

litura. In teak, it is one of about 139 defoliators that attack all stages from seedlings to mature trees (Roychoudhury et al., 1995). *S. litura* is abundant on teak in June and July and damage incidence in seedlings has been reported to be as high as 56%. Late-instar larvae were found to feed preferentially on mature teak leaves, whilst early instars fed on leaves of intermediate age. High concentrations of polyphenols in young leaves (Roychoudhury et al., 1995) may reduce their attractiveness to *S. litura* larvae but differing levels of susceptibility among nine teak clones were attributed to the nitrogen:potassium ratio of the foliage (Roychoudhury et al., 1998).

Studies elsewhere in southern Asia illustrate the economic impact of *S. litura*. In Pakistan, it is one of several lepidopteran pests attacking a wide range of crops including cotton and rice (Ahmad and Kamaluddin, 1987), as well as cabbage, tobacco, groundnut, soybean, alfalfa, gram, cowpea, tomato, cauliflower, carrot, onion, brinjal, turnip, radish and spinach (Maree et al., 1999).

Symptoms/Signs

On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants.

Cotton: Leaves are heavily attacked and bolls have large holes in them from which yellowish-green to dark-green larval excrement protrudes.

Tobacco: Leaves develop irregular, brownish-red patches and the stem base may be gnawed off.

Maize: The stems are often mined and young grains in the ear may be injured.

Known Hosts

The host range of *S. litura* covers at least 120 species. Among the main crop species attacked by *S. litura* in the tropics are *Colocasia esculenta*, cotton, flax, peanuts, jute, alfalfa, maize, rice, soybeans, tea, tobacco, vegetables, eggplants, *Brassica* spp., *Capsicum* spp., cucurbit vegetables, *Phaseolus* spp., potatoes, sweet potatoes and *Vigna* spp. Other hosts include ornamentals, wild plants, weeds and shade trees (for example, *Leucaena leucocephala,* the shade tree of cocoa plantations in Indonesia).

Both *S. litura* and *S. littoralis* are widely polyphagous (Brown and Dewhurst, 1975; Holloway, 1989).

Major Hosts

Abelmoschus esculentus (okra), *Acacia mangium* (brown salwood), *Allium cepa* (onion), *Amaranthus* (grain amaranth), *Arachis hypogaea* (groundnut), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Boehmeria nivea* (ramie), *Brassica*, *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Camellia sinensis* (tea), *Capsicum frutescens* (chilli), *Cicer arietinum*

(chickpea), *Citrus*, *Coffea* (coffee), *Colocasia esculenta* (taro), *Corchorus* (jutes), *Corchorus olitorius* (jute), *Coriandrum sativum* (coriander), *Crotalaria juncea* (sunn hemp), *Cynara scolymus* (artichoke), *Fabaceae* (leguminous plants), *Foeniculum vulgare* (fennel), *Fragaria ananassa* (strawberry), *Gladiolus* hybrids (gladiola), *Glycine max* (soybean), *Gossypium* (cotton), *Gossypium hirsutum* (Bourbon cotton), *Helianthus annuus* (sunflower), *Hevea brasiliensis* (rubber), *Ipomoea batatas* (sweet potato), *Jatropha curcas* (Barbados nut), *Lathyrus odoratus* (sweet pea), *Lilium* spp. (lily), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Malus domestica* (apple), *Manihot esculenta* (cassava), *Medicago sativa* (alfalfa), *Morus alba* (mora), *Musa* spp. (banana), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Papaver* (poppies), *Paulownia tomentosa* (paulownia), *Phaseolus* (beans), *Piper nigrum* (black pepper), *Poaceae* (grasses), *Psophocarpus tetragonolobus* (winged bean), *Raphanus sativus* (radish), *Ricinus communis* (castor bean), *Rosa* (roses), *Sesbania grandiflora* (agati), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Syzygium aromaticum* (clove), *Tectona grandis* (teak), *Theobroma cacao* (cocoa), *Trigonella foenum-graecum* (fenugreek), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), *Vigna unguiculata* (cowpea), *Vitis vinifera* (grape), *Zea mays* (maize), and *Zinnia elegans* (Zinnia).

Known Distribution

The tobacco caterpillar, *S. litura*, is one of the most important insect pests of agricultural crops in the Asian tropics. This species is widely distributed throughout tropical and temperate Asia, Australasia and the Pacific Islands (Feaking, 1973; Kranz et al., 1977).

Potential Distribution Within the US

The pest is present in Hawaii, but is not recorded from the continental U.S.

Survey

The presence of newly hatched larvae can be detected by the 'scratch' marks they make on the leaf surface. The older larvae are night-feeders and are usually found in the soil around the base of plants during the day. They chew large areas of the leaf, and can, at high population densities, strip a crop of its leaves. In such cases, larvae migrate in large groups from one field to another in search of food.

Developments in pheromone technology have made it possible to monitor *S. litura* in the field, to improve on timing of plant protection measures within peanut IPM programs.

The identification of a male sex pheromone of *S. litura*, (ZE) 9,11-tetradecadienyl acetate and (ZE) 9,12-tetradecadienyl acetate by Youshima et al. (1974) has enabled effective monitoring of this species for several years. The basic work regarding trap design, height, longevity of the septa, and the potential role of this technology in peanut has been thoroughly studied at ICRISAT Center,

Hyderabad, India over the past decade. These studies have clearly indicated the migratory behavior of the species in different areas. At present, pheromone technology has given high priority in monitoring for timing of plant protection measures within peanut IPM programs. The studies on trap density in peanut situations indicated no significant differences in moth catches when there were four or more traps per hectare. No decline was noticed in moth catch with increase in trap density. This indirectly suggests a limited utility in mass trapping operations (Ranga Rao et al., 1989).

Key Diagnostics

S. litura can be easily confused with *S. littoralis* as in both cases adults and larvae are similar, and they can be distinguished only through examination of genitalia. On dissection of the genitalia, ductus and ostium bursae are the same length in female *S. littoralis,* different lengths in *S. litura*. The shape of the juxta in males is very characteristic, and the ornamentation of the aedeagus vesica is also diagnostic. The presence of newly hatched larvae can be detected by the 'scratch marks' they make on the leaf surface.

For more information on the morphological discrimination between the adult, pupal and larval stages of the two species, see Mochida (1973).

Stink Bugs

Acrosternum hilare
Nezara viridula
Euschistus servus
Halyomorpha halys

Scientific Names - Common Name(s)
Acrosternum hilare Say – Green stink bug
Nezara viridula Linnaeus – Southern green stink bug
Euschistus servus Say – Brown stink bug
Halyomorpha halys Stal – Brown marmorated stink bug

Type of Pest
Stink bugs

Taxonomic Position
Class: Insecta, **Order**: Heteroptera, **Family**: Pentatomidae

Reason for Inclusion in Manual

Pest Description
Adults: Flattened, shield-shaped, and with fully developed wings (Fig. 1). Adults are frequent flyers. Stink bugs can be recognized by their ovoid, triangular body shape, narrow head, five segmented antennae, and malodorous scent. They are usually either green or brown.

Eggs: Barrel-shaped and are laid in regular clusters on the undersides of leaves.

Immatures (nymphs): Resemble the adults (colors are widely variable), except their wings are not fully developed and are not reproductively mature.

Pest Importance
Stink bugs can greatly impact soybean production as they are primarily attracted to reproductive stages and prefer to feed on developing seeds. While feeding,

stink bugs inject digestive enzymes into seeds, and the resulting wound also provides a site for disease entry. Stink bug feeding thus reduces pod development and seed quality and renders beans more likely to deteriorate in storage. Effected seed can have reduced germination if used for planting. *Nezara viridula* is the most important of the stink bugs attacking soybeans, because it also attacks several other cultivated crops. Annual soybean losses to *N. viridula* have been estimated as high as $23.5 million in Georgia alone (McPherson and McPherson, 2000). Throughout North America, *A. hilare* is tied for second in importance among all insects attacking soybean pods and seeds.

Figure 1. Stinkbug adults from left to right: Southern green stink bug *Nezara viridula,* Photo courtesy of Russ Ottens, The Bugwood Network; Green stink bug *Acrosternum hilare*, Photo courtesy of Marlin Rice, Iowa State University; Brown stink bug *Euschistus servus,* Photo courtesy of Marlin Rice, Iowa State University; and Brown marmorated stink bug *Halyomorpha halys* Photo courtesy of David R. Lance, USDA, APHIS, PPQ.

Symptoms/Signs

Both adults and nymphs of stink bugs have piercing and sucking mouth parts that act like hypodermic needles to remove the plant's fluids. Although both feed primarily on developing seeds and green pods of soybean, they also feed on plant stems, foliage, and blooms. Usually the location of feeding punctures can be recognized by the presence of small brown or black spots. Damaged pods have puncture marks surrounded by a darkened area of dead tissue. Young seeds can be deformed, undersized or even aborted, whereas older seeds are discolored and shriveled. Feeding damage can delay plant maturity and cause the abnormal production of leaflets and pods, a condition referred to as the 'green bean effect'. Feeding by *A. hilare, E. servus, and H. halys* also causes 'cat-facing' scars on the fruit, making it unmarketable.

Known Hosts

Stink bugs feed on a wide range of cultivated and wild host plants. Cultivated hosts include: soybean, snapbeans, corn, cotton, clover, peas, okra, lima beans, peaches, and alfalfa. *Halyomorpha halys* have a particular affinity for Fuji apples, as reported from Japan, China and South Korea. Stink bugs also feed on a large number of wild hosts including: black locust, elderberry, honey locust, wild cherry, mimosa, redbud, dogwood, box elder, silver maple, Norway maple, European linden (bass-wood), American linden, wild cherry, elderberry, black-haw, peach, apple, catalpa, and grasses.

Known Distribution

Acrosternum hilare ranges from Quebec and New England west through southern Canada and the northern United States to the Pacific Coast and south and southwest to Florida, Texas, Arizona, Utah, and California (McPherson 1982). *Nezara viridula* is believed to have originated in Ethiopia and spread to Europe, Asia, Africa and North and South America.

Potential Distribution Within the US

In the United States, *A. hilare* replaces *N. viridula* farther north and the two species are joined by the *E. servus* complex in the south (McPherson and McPherson, 2000). *Euschistus servus* is more wide ranging and occurs throughout the North America, comprising two subspecies, *E.* subsp. *servus*, that occurs from the southeastern U.S. west through Louisiana, Texas, New Mexico, and Arizona into California; and *E.* subsp. *euschistoides*, that occurs across Canada and the northern part of the U.S. (McPherson, 1982). The two subspecies intergrade in a broad band from Maryland to Kansas (Sailer, 1954). *Halyomorpha halys* is indigenous to Asia and was recently recorded for the first time in the U.S. in Allentown, Pennsylvania.

Survey

Adults and late instar nymphs can be sampled using sweep nets or drop cloths. Several sites in a given field should be sampled because of the insect's clumped distribution. Females are highly attracted to the soybean plant during bloom stage, so fields in flower should be sampled first. Counts of later-instar nymphs and adults can be combined for purposes of management decisions. Soybeans should be checked periodically for stink bug injury throughout pod development.

Key Diagnostics

Stink bugs can be recognized by their ovoid, triangular body shape, narrow head, five segmented antennae, and malodorous scent. They are usually either green or brown. Usually the location of feeding punctures can be recognized by the presence of small brown or black spots. Damaged pods have puncture marks surrounded by a darkened area of dead tissue.

Thrips

Frankliniella intonsa

Scientific Name
Frankliniella intonsa Trybom

Synonyms:
Frankliniella breviceps, Frankliniella brevistylis, Frankliniella formosae f.sp. *tricolor, Frankliniella intonsa* f.sp. *norashensis, Frankliniella intonsa* var. *Maritima, Frankliniella intonsa* var. *Rufula, Frankliniella vicina, Physopus brevistylis, Physopus vulgatissimus, Physopus vulgatissimus* var. *Adusta, Physopus vulgatissimus* var. *Albicornis, Physopus vulgatissimus* var. *Fulvicornis, Physopus vulgatissimus* var. *Nigropilosa, Thrips intonsa, Frankliniella formosae*

Common Name(s)
Taiwan flower thrips, flower thrips

Type of Pest
Thrips

Taxonomic position:
Class: Insecta, **Order:** Thysanoptera, **Family:** Thripidae

Reason for inclusion in manual

Pest Description
First larval instar: Posterior margin of abdominal segment 9 with about 12 narrowly cuneate processes dorsally and laterally, each approximately 4 µm long and arranged at regular intervals. Integument dorsally with indistinct oval plaques devoid of microtrichia; from mesothorax to abdominal segment 8 dorsally with minute circular plaques each bearing a microtrichium. Integument of thoracic segments and first segment ventrally with small oblong plaques devoid of microtrichia; abdominal segment 2 to 8 as in dorsal aspect. Setae with 'knobbed'-apices are D1 and D2 of abdominal segment 9 and D1 on segment 10. The other setae are pointed. The longest setae on the meso- and metathoracic coxae are 25 to 30 µm long. (Speyer and Parr, 1941).

Second larval instar: Orange-yellow. Posterior margin of abdominal segment 9 with a yellowish-grey band dorsally, which includes the median setae insertions. Posterior half of abdominal segment 10 yellowish-grey. Posterior margin of abdominal segment 9 dorsally and laterally has 18 narrowly cuneate processes, each about 4 µm long, arranged at irregular intervals; laterally these processes are longer. Mesothorax up to abdominal segment 13 has transverse rows of small lightly raised rounded or oval plaques, the majority of which bear a short microtrichium; upon the abdominal segment 7 and 8, the microtrichia become longer; thorax and first abdominal segment has rather more prominent oval or oblong plaques devoid of microtrichia dorsally; abdominal segment 2 to 8 ventrally as in dorsal aspect. Abdominal segment 2 and 8 with small peritremes, about 10 µm in diameter. All setae are pointed (Speyer and Parr, 1941).

Adult female: Head wider than long, widest at base; faintly striate posteriorly; interocellar setae developed, placed within the ocellar triangle. Fourth postocular setae pair most developed, whereas first to third pairs are small. Antennal segments 1 and 2 are brown, second darker; 3 to 5 yellowish; apices of segments 4 and 5 brownish; 6 to 8 brown; segments 3 and 4 each with forked sensory cone. Mouth-cone narrowly rounded in shape. Pronotum nearly smooth, major setae developed; anteroangular setae slightly longer than anteromarginals; posteroangular setae with inner pair longer than other pair. Legs predominantly yellow; femora, mid and hind tibiae darkened medially; hind tibiae with row of spine-like setae on inner margin. Forewings clear yellow; vein setae, strong, dark; posterior fringe cilia wavy; scale yellow. Mesonotum with faint, widely spaced transverse striae. Metascutum transverse reticulate medially; longitudinally striate laterally; median setae long, placed near anterior margin. Abdominal tergites with weak lateral setae developed, placed far apart. Tergite 8 with sparse comb of microtrichia on posterior margin. Tergites 9 and 10 with long, dark apical setae, pointed at apex. Sternites without accessory setae; sternite 6 with inner pair of posteromarginal setae placed anterior of posterior margin (Reyes, 1994).

Adult male: Similar in structure to females. Head yellow in color; thorax orange-yellow; abdomen grey; antennal segments 1 to 5 yellow; apices of segments 4 and 5 brown; segments 6 to 8 brown. Abdominal tergite 9 with B1 setae pale, moderately stout, pointed at apex; lateral setae dark, stout and pointed at apex. Abdominal sternites 3 to 7 each with a

Figure 1. Adult *F. intonsa*. Photo courtesy of Maria Pobożniak, Department of Plant Protection, Agricultural University of Cracow, Poland.

transverse glandular area (Reyes, 1994).

Pest Importance

In only a few crops has economic damage been ascribed specifically to the occurrence of *Frankliniella intonsa*: asparagus (Tang, 1975), chrysanthemum (Wang, 1982), okra (Toyota, 1972), tomatoes (Toyota, 1972; Murai, 1988) and peas (Wang, 1990; Fang, 1996). As part of a pest complex, *F. intonsa* has been associated with economic damage to strawberries in Italy and the United Kingdom (Buxton and Easterbrook, 1988; Gremo et al., 1997), alfalfa in former Czechoslovakia (Rotrekl, 1985) and nectarines in Greece (Kourmadas et al., 1982).

Symptoms/Signs

F. intonsa causes direct damage by feeding (suction injury) and egg laying in fruits. In mixed thrips populations, it is almost impossible to determine the specific damage caused by *F. intonsa*, although the following symptoms have been recorded: skin 'russeting' of nectarines; diminished fruit set in alfalfa; and distorted strawberry fruits (Kourmadas et al., 1982; Rotrekl, 1985; Buxton and Easterbrook, 1988; Gremo et al., 1997). In Taiwan and Japan, *F. intonsa* is known to damage different stages of pea (Fang, 1996; Kakizaki, 1996). Hachiya (1990) determined that on adzuki bean (*Vigna angularis*), almost 10 adults were required per flower to cause visible damage (CABI, 2004)

In Japan, the occurrence of white swellings and spots on tomato fruit caused by oviposition by *F. intonsa* was confirmed (Murai, 1988). This injury was recorded all over Japan, and its occurrence was greater than 30% in some regions.

Indirect damage is caused by the transmission of tospoviruses. Symptoms caused by tospovirus transmission are described by Loebenstein et al. (1995).

Known Hosts

F. intonsa has been recorded from many different plant species. Miyazaki and Kudo (1988) present a list of 146 species recorded as host plants and other records add many other plants on which the thrips have been found. Some of the plants mentioned in these lists are probably not true hosts, however, as thrips reproduction on the host was not always established (Mound and Teulon, 1995). *F. intonsa* normally occurs together with other Thripidae in flowers, necessitating identification of individual larvae to establish evidence of reproductive hosts. With respect to reproduction, *F. intonsa* is highly dependent on pollen (Murai and Ishii, 1982).

Major hosts

Abelmoschus esculentus (okra), *Arachis hypogaea* (groundnut), *Asparagus officinalis* (asparagus), *Capsicum annuum* (bell pepper), *Chrysanthemum indicum* (chrysanthemum), *Fragaria* spp. (strawberry), *Glycine max* (soybean), *Gossypium* (cotton), *Lycopersicon esculentum* (tomato), *Medicago sativa*

(alfalfa), *Oryza sativa* (rice), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Prunus persica* (peach), *Rosa* spp. (rose), and *Vigna angularis* (adzuki bean)

Known Distribution

In Asia, *F. intonsa* is easily transported by the international cut flower trade. Many consignments of cut chrysanthemum flowers grown in Taiwan for export to Japan fail quarantine examinations, in part due to the presence of *F. intonsa* (Wang, 1982). In the Netherlands, the number of interceptions of *F. intonsa* is far less than those of *F. occidentalis* or *F. schultzei* (Vierbergen, 1992). During the summer, *F. intonsa* is known to occur in greenhouses in Europe (Vierbergen, 1988; Sauer, 1997), but unlike most greenhouse insects, spread to other countries with intensive greenhouse culture has not been reported.

Potential Distribution Within the US

F. intonsa is present in Washington State and Oregon.

Survey

Flower thrips, like *F. intonsa,* are most commonly found inside flowers, feeding mostly on pollen. Several collection methods can be used for detection, collecting and conservation of larvae, including removal with a moist paintbrush, and beating from affected plant parts onto a suitably colored board or a white cloth (fixed in a frame). Dragging vegetation with fine-mesh nets collects both larvae and adults. For collecting thrips, specimens can be preserved initially in a tube with ethyl alcohol and a small amount of glycerine (Mantel and Vierbergen, 1996).

Plant parts can be enclosed in a plastic bag, in which a piece of filter paper is enclosed to absorb condensation. After 24 hours, most thrips leave the plant material and can be found on the inside of the closed bag.

Affected plant material, such as ears, turf, fallen leaves, moss and dead branches of trees can be processed in a Berlese apparatus after the method of Löser and Wetzel (1983). Modifying the apparatus by using an electric light bulb and closed lid resulted in most thrips being collected within 48 hours, due to heat and drying of the plant material.

Winged adults can be caught using yellow or blue sticky boards (Fang, 1993; Song et al., 1997). For microscopic research the boards have to be covered with transparent plastic. The specimen, together with a piece of the plastic and a small amount of glue can then be removed and placed in dichlormethan. After dissolving of the glue, the specimen is then ready for preparation (CABI, 2004)

Key Diagnostics

Microscopic slide preparation is required for accurate identification. Non-permanent slides can be made using lactic acid or Hoyer's solution, whereas permanent preparations can be made using Canada balsam (CABI, 2004).

Frankliniella is a large genus that includes about 150 species. Moulton (1948) reviewed the genus and provided keys for the identification of *Frankliniella* species known at the time, including *F. intonsa.* The species most commonly confused with *F. intonsa* is the cosmopolitan western flower thrips (*F. occidentalis*). Adults of *F. intonsa* can be distinguished from *F. occidentalis* by the length of postocular seta i3, which in *F. occidentalis* is more (and *F. intonsa* less) than three times the length of each of the other postocular setae. The metanotal campaniform sensillae are always missing in *F. intonsa* and are normally present in *F. occidentalis*.

Thrips palmi

Scientific Name
Thrips palmi Karny

Synonyms:
Chloethrips aureus, Thrips clarus, Thrips gossypicola, Thrips gracilis, Thrips leucadophilus, Thrips nilgiriensis

Common Name(s)
Melon thrips, oriental thrips, southern yellow thrips

Type of Pest
Thrips

Taxonomic Position
Class: Insecta, **Order:** Thysanoptera, **Family:** Thripidae

Reason for Inclusion in Manual

Pest Description
Adults**:** Thysanoptera adults can usually be distinguished from other insects by their slender wings which are fringed with long hairs. However, the adults of many species are wingless as are the young stages. Adults also differ from other insects in having an eversible bladder on each tarsus, and only have a single mandible in their heads. Two suborders are recognized; in one the females have a saw-like ovipositor, whereas in the other, both sexes have the last abdominal segment tubular.

Figure 1. Adult *T. palmi* Female. Photo courtesy of Yu Yan-Fen (CABI, 2004).

The structure of the mouthparts of adult females of *Thrips palmi* have been examined by Yasumi et al. (1994); the morphology of mouthparts, and the feeding marks on injured leaves indicate that *T. palmi* is a sap feeder.

Female: Color pale yellow (Figs. 1, 2), except antennal segment III usually dark

at apex, IV and V usually dark with base pale, VI and VII dark; forewings pale. Antennae with seven segments, terminal segment small; segments III and IV with forked sense cones. Head with no setae directly in front of first ocellus, one pair lateral to first ocellus and a smaller pair nearer the compound eyes; postocellar and postocular setae small. Pronotum with two pairs of long posteroangular setae, remaining setae small; surface with faint transverse lines. Metanotum with median pair of setae not at anterior margin, sculpture converging to posterior, paired companiform sensilla present. Forewing first vein with only 2 or 3 setae distally but with about 7 setae basally; second vein with a row of about 12 setae. Abdominal tergite VIII posterior margin with a comb of long, fine microtrichia, paired ctenidia present posteromedially from the spiracles; tergite IX with two pairs of campaniform sensilla; tergite II with 4 lateral setae; median tergites with median setae shorter than the distance between their bases, and no sculpture medially, lateral sculpture without microtrichia. Abdominal sternites with 3 pairs of posteromarginal setae, but no discal setae; pleurotergites lacking microtrichia and discal setae**.**

Figure 2. Adult *T. palmi*. Photo courtesy of Zenkoko Noson, Kyoiku Kyoiku Co. Ltd, Japan

Male**:** Similar to female but smaller; tergite II sometimes with only 3 lateral setae; tergite VIII posteromarginal comb often absent laterally; tergite IX setae B1 slightly shorter than, but in line with B2 setae; sternites III to VII each with a large transverse glandular area.

Larvae**:** In common with other, similar thrips species, *T. palmi* has two larval stages and two pupal stages. The second-instar larvae (Fig. 3) can be distinguished from those of other species by details of the sculpture of the dorsal surface, but specimens are even more difficult to prepare for study than are adults. Miyazaki and Kudo (1986) provide an identification key to several species, which are common in the Oriental region.

Pest Importance

Walker (1992, 1994) has reviewed the pest status of *T. palmi*; much of the information given here is from these reviews, together with updated information the last 5 years of published information. The importance of the pest on vegetable crops in Southeast Asia was emphasized by a workshop held in Bangkok, Thailand (Talekar, 1991) where seven of the eight papers presented listed *T. palmi* as causing concern for vegetable growers in their region.

Figure 3. Second-instar larvae. Photo courtesy of Y. Hirose (CABI, 2004).

The economic injury density in Japan has been estimated at 0.105 adults per flower or 4.4 adults

per sticky trap per day on *Capsicum annuum* in greenhouses (Morishita and Azuma, 1988), assuming an acceptable yield loss of 5% of the maximum yield. Kawai (1986b) also reported that economic injury thresholds were low in greenhouses in Japan, assuming an acceptable yield loss of 5% of the maximum yield, with 0.08 and 4.4 adults per leaf for eggplant and cucumber, respectively, and 0.11 adults per flower for *C. annuum*. Morishita and Azuma (1989) considered counting injured fruits to be a better sampling method than counting insects on leaves.

Medina (1980) reported that *T. palmi* had been found in the Philippines as early as 1977, and that this outbreak destroyed almost 80% of the watermelon plantations in central Luzon and Laguna. Plantings of eggplant intended for seed production had to be abandoned due to severe *T. palmi* damage and even the application of insecticide as often as every 4 days failed to provide satisfactory control (Bernardo, 1991). Chang (1991) lists *T. palmi* as one of Taiwan's most important pest thrips; damage was first observed on cucurbits in 1979, but the species was incorrectly identified as *T. flavus. T. palmi* has also been identified as an important pest of potato in Taiwan by SEAMEO SEARCA (1991).

However, Bournier (1986) reported that *T. palmi* caused insignificant damage on cotton, tobacco and wild plants in Java, Sumatra and India. Miyazaki et al. (1984) also observed, during a survey of soybean in Java, that *T. palmi* did not cause heavy damage except in one instance on eggplant.

Cooper (1991b) recorded infestations of 300 to 700 *T. palmi* per leaf on eggplant and cucumber, resulting in crop losses of 50 to 90% in Trinidad. He suggested that *T. palmi* may have been brought to Trinidad in the winds of a tropical depression during 1988, but it has also been postulated that it may have gained entry through plant material from another Caribbean island, for example Martinique, where it is reported as a serious pest. Pantoja et al. (1988) noted that the climatic conditions in Puerto Rico are favorable for the early development of large populations of *T. palmi* on commercial crops, as well as on weeds. Guyot (1988) reported the disastrous economic effect that *T. palmi* had in Guadeloupe when eggplant exports fell from 5000 tons in 1985 to 1600 tons in 1986, and in Martinique where 37% of the vegetable crops of the two main co-operatives were attacked by *T. palmi*, including 90% of eggplant crops.

In Hawaii, Johnson et al. (1989) observed that, together with *Aphis gossypii*, *T. palmi* was the major foliar pest on Oahu (1984-85). Welter et al. (1989) studied mixed infestations of *T. palmi* and the western flower thrips, *Frankliniella occidentalis*, and noted significant reductions in total cucumber yield, mean fruit size and total fruit. The population trends of *T. palmi* on commercial watermelon plantings in Hawaii were surveyed by Johnson (1986). Peak infestation levels varied from 2.5 to 53.6 individuals per leaf and from 18 to 97% infested vine tips per planting.

Johnson (1986) pointed out that *T. palmi* could establish itself in the continental U.S., given the extensive flow of air traffic between Hawaii and the mainland, especially California, but it was not until 1991 that *T. palmi* was found in the U.S., not in California as predicted by Johnson but in Florida (FAO, 1991). Heavy infestations were detected on potato, eggplant, *Capsicum* spp., *Phaseolus vulgaris*, yellow squash and several weeds.

Symptoms/Signs

Damage by *T. palmi* is not unlike that caused by many other species of thrips; when populations are high, their feeding causes a silvery or bronzed appearance on the surface of the plant (Fig. 4, 5), especially on the midrib and veins of leaves and on the surface of fruit. Leaves and terminal shoots become stunted and fruit is scarred and deformed. Damaged leaves generally show a darkened, glossy, pearly appearance (Bournier, 1987). Johnson (1986) described heavy damage to watermelon foliage as bronzing and total destruction of the vine tips.

Bournier (1983, 1987) described damage to cultivated cotton caused by *T. palmi* and,

Figure 4. Bean leaf bronzing caused by an infestation of melon thrips, *Thrips palmi*.. Photo courtesy of John Capinera, University of Florida.

among the symptoms, observed that the oldest tissue may thicken, warp and finally crackle. Damage to cotton seedlings by *T. palmi* has also been reported in Thailand by Wangboonkong (1981) if there are long periods of drought early in the season.

Apart from Karny (1925), who described *T. palmi* and observed that it infested

both mature and seedling tobacco in Sumatra, one of the earliest reports of damage by *T. palmi* was Ananthakrishnan (1955). He described the damage to Sesamum plants in Madras, India, as malformation of the stamens, injury to the ovarian wall and the development of a dark pigment on the fruit wall, instead of the usual green color.

Damage has been described by Nakazawa (1981) in Japan as yellowing of the

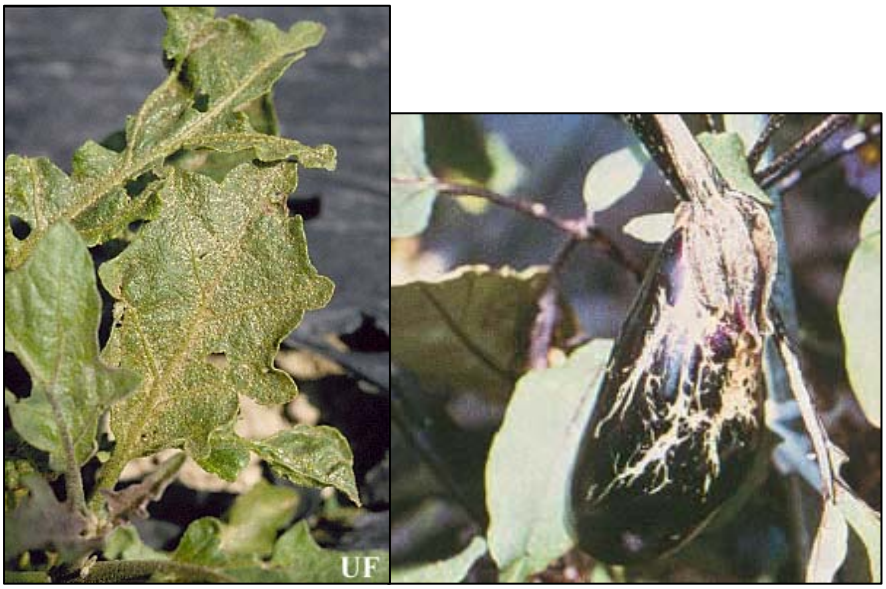


Figure 5. Eggplant damage caused by melon thrips (left), Photo courtesy of John Capinera, University of Florida; Eggplant scaring damage caused by melon thrips (right), Photo courtesy of FDACS – Division of Plant Industry.

leaves, topping, scratches on the fruits, malformation of the fruits, poor fruiting and death of the whole plant when populations are high. In Martinique, Denoyes et al. (1986) described the damage on the leaves of eggplant, cucumber, melon and other cucurbits. Pantoja et al. (1988) reported severe damage to cucurbits and solanaceous commercial plantings in 1986 in Puerto Rico, where adult and immature thrips fed gregariously on leaves, stems, flowers and developing fruits. Pepper plants became stunted with a bronzed appearance and eggplant plants showed premature fall of developing fruits and buds, and deformed fruits.

Kawai (1986b) studied the relationship between the density of *T. palmi* and the damage to *Capsicum annuum* and eggplant in Japan. He also studied the relationship between different densities of *T. palmi* and injury to cucumbers grown in a greenhouse (Kawai, 1986c). The growth of cucumber plants was retarded when thrip numbers were high. The tolerable pest densities were estimated at 5.3 adults per leaf for the total fruit yield and 4.4 adults per leaf for the yield of uninjured fruit (assuming an acceptable yield loss of 5% of the

maximum yield).

Sakimura et al. (1986) observed that both adults and larvae of *T. palmi* feed gregariously on leaves, firstly along the midribs and veins. Stems are attacked, particularly at or near the growing tip, and are found amongst the petals and developing ovaries in flowers and on the surface of fruit. They leave numerous scars and deformities, and finally kill the entire plant.

Known Hosts

Major hosts

Allium cepa (onion), *Capsicum* (peppers), *Capsicum annuum* (bell pepper), *Chrysanthemum* (daisy), *Citrus* spp., *Cucumis melo* (melon), *Cucumis sativus* (cucumber), *Cucurbita pepo* (ornamental gourd), *Cucurbitaceae* (cucurbits), *Fabaceae* (leguminous plants), *Glycine max* (soybean), *Gossypium* spp.(cotton), *Helianthus annuus* (sunflower), *Lactuca sativa* (lettuce), *Lycopersicon esculentum* (tomato), *Mangifera indica* (mango), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Persea americana* (avocado), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Sesamum indicum* (sesame), *Solanaceae*, *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), and *Vigna unguiculata* (cowpea)

Known Distribution

Over the past 10 years *T. palmi* has rapidly become a major pest of cucurbits and solanaceous plants and has gained a foothold in many tropical regions of the world.

The oldest voucher specimens of *T. palmi* in the collections of the Natural History Museum, London are from Thailand in 1947, India (West Bengal) in 1950, Pakistan (North West Himalaya) in 1951, Malaysia in 1971, and the Philippines in 1977; most of the available material is from the 1980s. This suggests that *T. palmi*, although widespread for many years, has both extended its range and recently become much more aggressive as a pest. Bournier (1986, 1987) considered that *T. palmi* may have been transported by wind to various Pacific islands, and also by aircraft to Japan on imported greenhouse plants.

Since its discovery in Japan in 1978, it has invaded a number of Pacific Islands, including Hawaii, and has been reported from northern Australia (Layland, 1991). It appears to be still spreading in the Caribbean since it was first discovered in 1985 and it has reached the southern U.S. (Florida) and tropical America (Guyana), Brazil and Argentina. *T. palmi* has established in Mauritius and Reunion, and there is one report from the Canary Islands, so it is probably only a matter of time before it is widely established in Africa. It is already present in Sudan. Moreover, it has now invaded greenhouses in temperate regions of the world and has been intercepted on cut flowers imported into Finland.

In Japan, Nakazawa (1981) reported that *T. palmi* had first been recognized in

1978 at Miyazaki on Kyushu Island. In 1982, Nakazawa drew attention to the fact that *T. palmi* was rapidly extending its range. Since then, various authors have reported extensive annual outbreaks severely affecting year-round plantings in many vinyl-covered warmhouses in Okinawa and the warmer coastal strips of Kyushu and Shikoku, and further north in central Japan (Sakimura et al., 1986). Outdoor overwintering normally occurs in Okinawa (26°N), but in the southern part of Kyushu (about 32°N) and further north, there is no overwintering outdoors and greenhouses serve as foci of summer populations.

Potential Distribution within the US

The pest is present in Florida and Hawaii.

Survey

T. palmi is not easily detectable because of its small size, so quarantine procedures are difficult to manage and this pest has probably slipped through the net with increased traffic in plant produce around the world.

Sakimura et al. (1986) observed that both adults and larvae of *T. palmi* feed gregariously on leaves, firstly along the midribs and veins. Stems are attacked, particularly at or near the growing tip, and they are found amongst the petals and developing ovaries in flowers and on the surface of fruit. They leave numerous scars and deformities, and finally kill the entire plant. Ho et al. (1993) reported that old eggplant leaves are a good site for sampling in Taiwan.

Layland et al. (1994) described methods used to monitor *T. palmi* using blue sticky-board traps and water-tray traps in Northern Territory, Australia.

Kawai (1983b) explored the relationship between the density of adults on cucumber plants and the number of individuals trapped by sticky traps in a plastic greenhouse in Japan. A positive correlation was found between the adult density per plant and the number of adults trapped. He concluded that adhesive traps can be used to monitor the relative abundance of *T. palmi* adults. Kawai (1983b) tested the attractiveness of blue and white for *T. palmi*. Huang (1989) observed that white was the most effective color to attract *T. palmi* to sticky trap plates and that 0.5 meters above ground level was the most suitable height to trap thrips. Layland et al. (1994) used blue sticky-board traps to monitor *T. palmi*.

Key Diagnostics

T. palmi can easily be confused in the field with several commonly found small, yellow species of thrips, such as *T. flavus*, or the pale forms of *T. tabaci*, *Frankliniella schultzei*, *F. occidentalis* and *T. nigropilosus*.

T. flavus is generally larger than *T. palmi*, whereas the yellow species of the common genus *Scirtothrips* are smaller. Moreover, the other species mentioned here nearly always have some brown shading on the abdomen, and *T. tabaci* is unusual in lacking red pigment beneath the ocelli.

A key to the species of adult female terebrantian thrips found on citrus flowers and floral buds in Florida is provided by Childers and Beshear (1992). Layland et al. (1994) describe techniques for rapid preparation and identification, with a brief description of specific characters, which may be used to differentiate *T. palmi* from four other closely related, economically important species found in Australia (*T. imaginis*, *T. tabaci*, *T. hawaiiensis* and *T. simplex*).

Zur Strassen (1989) stressed the likelihood of *T. palmi* being introduced into Europe, described *T. palmi*, and gave the characters which may be used to distinguish it from several other species in Europe.

Diseases

Bacterial Diseases

Pseudomonas savastanoi pv. *glycinea*

Scientific Name
Pseudomonas savastanoi pv. *glycinea* Coerper, Gardan et al., 1992

Synonyms:
Pseudomonas syringae pv. *glycinea*, *Pseudomonas glycinea*, *Pseudomonas glycinea* var. *japonica*, *Pseudomonas syringae* pv. *glycinea*

Common Name(s)
Bacterial blight, bacterial blight of soybean

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Phylum: Proteobacteria, **Class**: Gamma Proteobacteria, **Order**: Pseudomonadales, **Family:** Pseudomonadaceae.

Reason for Inclusion in Manual

Notes on Nomenclature
Young et al. (1978) proposed a new nomenclature and classification for plant-pathogenic bacteria and introduced the epithet 'pathovar' for the infrasubspecific level. All fluorescent oxidase-negative *Pseudomonas* species (except *P. viridiflava*) were assigned to a single species, *Pseudomonas syringae*, containing several pathovars. *P. savastanoi*, which attacks plants of the family Oleaceae, was reclassified as *P. syringae* pv. *savastanoi*. Later, Jansen (1982) elevated the epithet *savastanoi* to a new subspecies, *Pseudomonas syringae* subs. *savastanoi*. Gardan et al. (1992) found that *Pseudomonas syringae* subsp. *savastanoi* strains belong to a DNA-related group including strains of *P. syringae*

pv. *phaseolicola* and *P*. *syringae* pv. *glycinea*. Thus, *Pseudomonas syringae* subs*. savastanoi* was elevated to species level as *P. savastanoi* pv. *glycinea*.

Pest Description

P. savastanoi pv. *glycinea* is an aerobic Gram-negative bacterium, 1.2 to 1.5 x 2.3 to 3.0 µm rods with rounded ends, and motile with one to several polar flagella. It develops, white and raised, circular, smooth, glistened, entire margin, and not viscid colonies on nutrient agar. The optimal temperature for growth is 24° to 26°C, but it can grow from 2° to 35°C. It weakly produces a yellow green, water soluble, fluorescent pigment in culture, and several toxins, responsible for some of the symptoms. The pathogen has several races, of which nine where found in the U.S. (Sinclair, 1999; Prom and Venette, 1997).

Biology and Ecology

P. savastanoi pv. *glycinea* survives in infected seeds and in soybean debris on the soil surface or buried, especially under dry and cold conditions (Park and Lim, 1985). Populations of the pathogen on seedlings from infected seeds may also be a significant source of inoculum for leaf infections (Daft and Leben, 1972). The pathogen enters the plant through stomata, multiplies in the intercellular spaces of the mesophyll, and produce toxins, which inhibit chlorophyll synthesis. Bacterial slime and fluids fill the intercellular spaces leading to the typical water-soaked lesions within 5 to 7 days. The bacterium is spread during windy rainstorms or cultivation while foliage is wet (Sinclair, 1999), and it can grow as epiphyte on buds and leaves until the proper temperature and moisture levels develop for infection (May et al., 1997).

P. savastanoi pv. *glycinea* can infect seeds through the pods during the growing season or during harvest. Its incidence in seeds may reach to >90%, and it can survive for 18 months or two years in seeds stored at 22°C or 3°C, respectively (Nicholson et al., 1973).

Bacterial blight may be controlled by the use of resistant cultivars. Most present cultivars have moderate levels of resistance. Four resistance genes, Rpg1, Rpg2, Rpg3, and Rpg4 have been identified. Planting of pathogen free-seeds, and crop rotation with non-susceptible crops to the pathogen are also recommended. Crop residues should be deep plowed, and cultivation during wet periods should be avoided.

Figure 1. Symptoms of bacterial blight. Photo courtesy of Clemson University-USDA Cooperative Extension Slide Series.

Pest importance

Bacterial blight is commonly found in cool and wet, temperate soybean-growing regions worldwide. The disease is most conspicuous during mid season under moist conditions, but it ceases to

develop during hot and dry weather. In the U.S., yield losses may range from 4 to 40% (Park and Lim, 1986; Williams and Nyvall, 1980); however, the disease does not cause significant economic losses if resistant or tolerant cultivars are used.

Symptoms/Signs

Symptoms of bacterial blight can be found on cotyledons, stems, petioles, leaves, pods and seeds. Symptoms are particularly conspicuous on leaves as small angular lesions, usually water-soaked at the center and surrounded by a yellow-green halo (Fig.1). Later, the spots coalesce to form dark-brown necrotic areas with yellow margins. Young leaves are the most susceptible and frequently show distortion, stunting and chlorosis. The spots are usually scattered over the leaf or grouped, resulting in irregular large necrotic areas. Later, the lesions become dry and fall out, giving the leaf a ragged appearance (Fig.2), especially after strong winds and thrashing rains (Sinclair, 1999).

On cotyledons, lesions develop first at the margins, these lesions enlarge, and turn dark brown as the tissue collapses. Seedlings from infected seed are stunted and die if the growing point is infected. On stems or petioles, lesions are blackish. On pods, lesions are initially small and water soaked, later they enlarge, merge to cover the whole pod, and turn dark brown to black with age. Infected seeds may be covered with a slimy bacterial growth. Stored seeds may shrivel, develop raised or sunken lesions, become slightly discolored, or appear healthy (Sinclair, 1999).

Figure 2. Symptoms of bacterial blight. Note the ragged appearance. Photo courtesy of X. B. Yang.

Known Hosts

The natural main host of *P. savastanoi* pv. *glycinea* is *Glycine max* (soybean). It can also affect *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (bush bean), *Phaseolus acutifolius* (tepary bean), *Vigna unguiculata* (cowpea), *Vigna angularis* (azuki bean), and *Pueraria lobata* (kudzu).

Known Distribution

P. savastanoi pv. *glycinea* has a worldwide distribution and occurs wherever soybeans are grown. The CABI (2004) list included: **Asia:** Brunei, China, India, Indonesia, Japan, Kazakhstan, Korea, Pakistan, and the Philippines. **Europe:** Austria, Bulgaria, France, Germany, Hungary, Italy, Moldova, Poland, Romania, Serbia and Montenegro, Ukraine, and the USSR. **Africa:** Egypt, South Africa, Zambia, and Zimbabwe. **North America:** Canada, Mexico and the U.S. **South**

America: Argentina, Brazil, Colombia, and Venezuela. **Oceania:** Australia and New Zealand.

Potential Distribution Within the US

Bacterial blight of soybean was originally described in Wisconsin in 1917 (Johnson and Coerper, 1917). Since then, it has been found in Alabama, Delaware, Florida, Hawaii, Illinois, Iowa, Minnesota, Missouri, Nebraska, North Carolina, North Dakota, Ohio, and Wisconsin. It is most likely present wherever soybean is grown.

Survey

Survey of bacterial blight in the field might be difficult because symptoms are very similar to other soybean diseases, and they are affected by the environmental conditions and plant growth stage. Samples for identification and isolation, should include both diseased and healthy plant tissue for comparison purposes. Dead leaves have little value. Samples should not be exposed to direct sunlight or high temperature; instead, they should be placed in plastic bags and stored in an ice chest to prevent drying.

Incidence of bacterial blight in fields may be assessed by systematic sampling in pre-selected patterns (W, X, triple diagonal, diamond). Leaves may be collected from specified sampling units (e.g., certain number of leaves can be collected from three 10 x 10 foot sample unit, 50 m apart, and 50 m from field edges). Prom and Venette (1997), in a survey for races collected 10 infected leaves in pairs from five plants arbitrarily selected along a W-shaped field sampling pattern.

Key Diagnostics

Small, angular, translucent, water soaked, yellow to light brown spots on leaves (Fig. 1). Center of lesions dried out, reddish brown to black, surrounded by a water-soaked margin, and bordered by a yellowish green halo. Large lesions drop out or tear giving the leaf a ragged appearance (Fig. 2). Symptoms are most evident on the leaves at the top of plants starting at V2 growth stage, especially with rain events and cool temperatures.

To identify *P. savastanoi* pv. *glycinea*, infected leaves can be macerated and the extract streaked onto King's B (KB) agar. Colonies of the pathogen produce a characteristic water-soluble, green-fluorescent pigment on KB agar (King et al., 1954). The bacterium is strictly aerobic and oxidase and arginine dihydrolase negative. It does not reduce nitrate, and does not accumulate poly-ß-hydroxybutyrate as a carbon-reserve material. The bacterium does not hydrolyze gelatin and arbutin. It produces acid but not gas from glucose and sucrose. The pathovar produces levan and can use the following carbon sources for growth: D-gluconate, m-inositol, mannitol, quinate, trigonelline, but not anthranilate, betaine, erythritol, DL-homoserine, DL-lactate, D-sorbitol, D(-)-tartrate, L(+)-tartrate

(Bradbury, 1986). *P. savastanoi* pv. *glycinea* is an efficient ethylene producer (Hildebrand, et al., 1988; Weingart and Volksch, 1997a).

P. savastanoi pv. *glycinea* can be distinguished from P. *syringae* pv. *tabaci* by the inability of the former to utilize betaine, sorbitol and erythritol, and is closely related to *P. savastanoi* pv. *phaseolicola*, which causes halo blight of bean. *P. savastanoi* pv. *phaseolicola* has an overlapping host range with pv. *glycinea*. However, pv. *phaseolicola* can not utilize inositol and mannitol, and cannot produce ethylene (Weingart and Völksch, 1997a). A good tool to distinguish these pathovars is the PCR method with ERIC, REP and IS50 primers (Weingart and Völksch, 1997b).

Pathogenecity of isolates can be tested by inoculating fifteen-day-old greenhouse-grown soybean seedlings (cultivars Oakland, Beeson, Acme and Flambeau) by rubbing leaves with a sterile cotton swab dipped in an aqueous suspension (~100,000 c.f.u./ml) of the presumptive pathogen. Inoculated seedlings and controls are incubated in light for 48 hr at 90% RH in a mist chamber at 25°C, then transferred to the greenhouse and observed for necrotic lesions on leaves 4 to 7 days after inoculation (Lelliot and Stead, 1987).

Alvarez et al. (1995) reported that Chauveau developed the technique widely used for seed certification. Five replicates of 100 g (±1000 seeds) seeds are soaked for 24 hours at 4 to 5°C in 600 ml of sterile tap water adjusted to pH 6.5 with a phosphate buffer solution. Three-fold serial dilutions are made from the soaking solution. Then, 0.1-ml aliquots are plated on KB medium amended with 10 µg/ml cephalexin (KBC). After incubation at 25°C for 2 to 3 days, presumptive colonies of *P. savastanoi* pv. *glycinea*, showing a blue fluorescence under UV light (370 nm), are re-isolated on KBC. Five presumptive colonies of each subsample are subcultured onto KB medium. These subcultures are then confirmed as *P. savastanoi* pv. *glycinea* by a positive reaction for levan production and negative reactions in oxidase and esculin hydrolysis tests (Hildebrand, et al., 1988).

A disease with similar symptoms is wildfire, which produces a distinct yellow halo with a necrotic center and is caused by *Pseudomonas syringae* pv. *tabaci*. The disease is more commonly found on tobacco.

Differentiation from Asian soybean rust

The disease can also be confused with Asian soybean rust. Bacterial blight develops angular spots surrounded by yellow halos. Asian soybean rust forms pustules on the underside of the leaf. Pustules have pores at the top of the cone and masses of uredionospores.

Xanthomonas axonopodis pv. *glycines*

Scientific Name
Xanthomonas axonopodis pv. *glycines* Nakano

Synonyms:
Xanthomonas campestris pv. *glycines*

Common Name(s)
Bacterial pustule, soybean bacterial pustule

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Phylum: Proteobacteria, **Class:** Gammaproteobacteria, **Order:** Xanthomonadales, **Family:** Xanthomonadaceae

Reason for Inclusion in the Manual

Notes on Nomenclature
Following the reclassification of *Xanthomonas* by Vauterin et al. (1995), *X. campestris* pv. *glycines* (Dye, 1978) was renamed as a pathovar in *X. axonopodis*, as *X. axonopodis* pv. *glycines*. Thus, *X. axonopodis* pv. *glycines* is now used as the preferred name.

Pest Description
X. axonopodis pv. *glycines* is an aerobic Gram-negative bacterium with rod shaped cells (0.5 to 0.9 x 1.4 to 2.3 µm), and single polar flagellum (Sinclair, 1999). It develops pale yellow, small, round, and smooth colonies with entire margins on beef infusion agar. It grows slowly in culture, and colonies become deep yellow with age. The optimum temperature for growth is 30 to 33ºC. Typically, *X. axonopodis* pv. *glycines* produces acid but not gas from sucrose, liquefies gelatin, and rapidly hydrolysis starch (Sinclair, 1999).

Biology and Ecology
Sources of primary inoculum are bacteria in infected seeds, crop residues, soil (Graham 1953; Groth and Braun, 1989), and in the rhizosphere of wheat roots

(Sinclair, 1999). Of these, seed transmission plays the most important role in disease development. *X. axonopodis* pv. *glycines* can survive on soybean seed for 2.5 years, on plant debris on the soil surface for 4 to 8 months, and in buried residues for 1 to 3 months (Feet, 1979). The pathogen can also survive in weeds such as *Brunnichia cirrhosa* in the U.S., *Macrotyloma uniflorum* in India, or in volunteer soybean plants. Secondary spread of the bacterium occurs in wet conditions via rain splash, agricultural implements, movement of animals and humans, foliage contact, and windblown rain. The disease spreads rapidly under rainy conditions (Graham, 1953; Allington and Feaster, 1946).

Figure 1. Symptoms of bacterial pustule. Photo Courtesy of A. Mengistu.

X. axonopodis pv. *glycines* enters the host plant through stomata or wounds, and invades the intercellular spaces, where it multiplies. Leaf cells at the infected site grow longer and multiply faster, in response to an extracellular compound secreted by the bacteria, which, in conjunction with the mass of bacteria, cause an epidermal expansion of both leaf surfaces. These raised areas rupture and become pustules in 5 to 7 days after infection. The disease is particularly prevalent in wet and warm (30 to 33°C) weather. New infections may develop anytime during the growing season, if conditions are favorable for the pathogen.

In the seeds, bacterial cells of *X. axonopodis* are localized in the hourglass layer of the seed coat, and in the intercellular spaces of palisade cells and sclerenchymatous cells of vascular bundles of the germinating embryo (Gupta and Pathak, 2002). Bacterial cells were also found in the parenchymatous cells just below the meristem.

Control of *X*. *axonopodis* pv. *glycines* include: use of resistant cultivars with *Rxp* gene; pathogen-free seeds; seed treatment (fungicides, hot water); deep plowing of diseased crop residues; and avoiding cultural practices during wet weather conditions. Other practices are rotation with non-legume crops and foliar sprays with copper alone or combined with antibiotics.

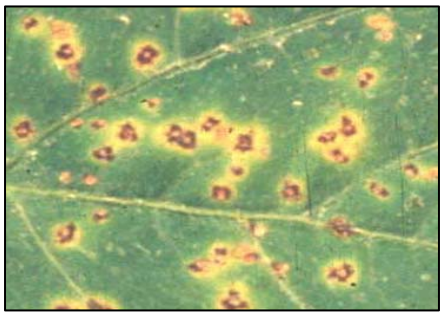


Figure 2. Close up symptoms of bacterial pustule. Photo Courtesy of G. Hartman.

Pest Importance

Bacterial pustule is moderately important economically, due to its worldwide distribution and seed transmission. It is the most prevalent bacterial disease of soybeans in Brazil, China, India, Korea, Sudan, U.S., and Taiwan. Bacterial pustule may cause premature defoliation, resulting in reduction in seed size

and number. Yield losses may reach 11% (Hartwig and Johnson, 1953).

Symptoms/Signs

Symptoms are generally confined to leaves. Small yellowish-green areas with reddish-brown and elevated pustule-like centers appear on one or both leaf surfaces (Figs. 1, 2). The raised centers or 'pustules' are more conspicuous on the lower leaf surface. Spots may merge, resulting in large irregular dead areas, which might be torn away by wind, giving the leaf a ragged appearance. Heavily infected leaves turn yellow and fall off. Susceptible plants may be completely defoliated. Sometimes, pustule-like lesions also develop on pods as small, reddish brown spots. Bacterial pustule may be differentiated from bacterial blight (*Pseudomonas savastanoi* pv. *glycinea*) by the presence of pustules and the absence of water-soaked areas, especially in the early stages of development.

Known Hosts

Major hosts

Glycine max (soybean) and *Phaseolus vulgaris* (common bean).

Minor hosts

Macrotyloma uniflorum (horsegram), *Phaseolus lunatus* (lima bean) and *Vigna unguiculata* (cowpea).

Wild hosts

Brunnichia cirrhosa (redvine).

Known Distribution

Bacterial pustule disease probably occurs in all the important soybean growing regions of the world with warm and moist climatic weather during the crop growing season. The CABI International (2004) list of countries where bacterial pustule has been found includes: **Asia:** Brunei, Cambodia, China, Taiwan, Georgia, India, Indonesia, Japan, Kazakhstan, Malaysia, Nepal, Philippines, and Thailand. **Europe:** Austria, Bulgaria, France, Lithuania, Moldova, Romania, USSR, Serbia and Montenegro, and theUkraine. **Africa:** Central African, Ivory Coast, Egypt, Ethiopia, Kenya, Madagascar, Malawi, Mozambique, Nigeria, Somalia, South Africa, Sudan, Tanzania, Uganda, Zambia, and Zimbabwe. **North America:** Canada, Mexico, and the U.S. **Central America:** Belize, Cuba, and Nicaragua. **South America:** Argentina, Bolivia, Brazil, and Venezuela.

Potential Distribution Within the US

In the U.S., bacterial pustule is present wherever soybean is grown.

Survey

The survey method suggested for bacterial blight in this manual may also be applicable for bacterial pustule. Symptoms of bacterial pustule are easily visible in the field after rainstorms or hailstorms. If lesions are not clear or where

confirmation of infection is required, leaves with suspected lesions should be incubated to induce bacterial multiplication.

Leaves: Look for bacterial pustule symptoms on leaves at the top of the plant. Lesions are small yellow-green spots with angular reddish-brown centers (Figs. 1 and 2). Spots have small, raised pustule-like structures in the center, especially those on the lower leaf surface. In contrast to bacterial blight, the water soaking is seldom seen.
Pods: Small, reddish-brown, slightly raised spots (pustules).

Key Diagnostics

The culture broth technique can be used to isolate *X*. *axonopodis* pv. *glycine*s from seed lots (Fett, 1979). Untreated or surface sterilized seeds (5 min. in 70% ethanol) are washed twice in sterile water, and shaken in 65 ml of nutrient broth for 18 to 24 h at 24°C. The broth culture is used to inoculate susceptible soybean plants.

Srivastava and Singh (1986) reported that plating seeds on water agar is more effective than the standard blotter method. Untreated seeds are incubated on water agar plates at 25°C for 72 hours and examined for bacterial colonies. Germinated seedlings are examined for black and circular slightly raised spots, after 10 days of plating. Identity of isolates from colonies or lesions must be confirmed by pathogenicity and biochemical tests.

Tests for identification of *X*. *axonopodis* pv. *glycines* include among others, gelatin hydrolysis, catalase production, salt tolerance, lypolitic activity, levan formation, starch hydrolysis, carbon utilization (Schaad et al., 2001) and growth on the semiselective MXG medium (Prathuangwong et al., 1997). For the pathogenicity test, suspected isolates are grown at 28ºC for 48 hours on NGA, then calibrated with sterile distilled water to ~1.0 x 10^8 CFU mL^{-1}. The bacterial suspensions are supplemented with 5 grams/liter of 600 mesh carborundum and 0.25 ml/liter Triton X 100, and sprayed onto the foliage of soybean plants (Kaewnum et al., 2005). Plants are observed 3 to 7 days after inoculation and evaluated for the disease severity (number of pustules per 1 cm^2 section of leaf).

Lazo and Gabriel (1987) found that, in most cases, strains of *Xanthomonas campestris* can be differentiated at the pathovar level on the basis of their characteristic plasmid DNA sequences. Thus, *X*. *axonopodis* pv. *glycines* (*X*. *campestris* pv. *glycines*), *X*. *campestris* pv. *malvacearum*, *X*. *campestris* pv. *phaseoli* and *X*. *campestris* pv. *vignicola* were accurately identified through determination of the restriction fragment profile and/or by Southern hybridizations of that profile.

X. *axonopodis* pv. *glycines* (*X*. *campestris* pv. *glycines*) produces the bacteriocin glycinecinA, against most xanthomonads. Oh et al. (1999) showed that a 1.7 kb DNA region for the glycinecinA gene was specific for *X*. *campestris* pv. *glycines*.

Therefore, the 1.7 kb DNA region for the glycinecinA gene can be used for the pathovar-specific probe for DNA hybridization and the primers heu2 and heu4 which amplify a segment within the 1.7 Kb DNA region can be used as pathovar-specific primers for PCR analysis to detect *X*. *axonopodis* pv. *glycines*.

Differentiation from Asian soybean rust

Pustules of Asian soybean rust have pores at the top of the cone and masses of uredionospores, whereas, bacterial pustules have fissures rather than pores, and the presence of the bacterium can be tested by the ooze test.

Fungal Diseases

Cercospora kikuchii

Scientific Name
Cercospora kikuchii, Matsumato & Tomoyasu, M. W. Gardner

Synonyms:
Cercosporina kikuchii

Common Name(s)
Purple seed stain, Cercospora blight, Cercospora leaf spot, lavender spot, purple patch, purple speck, purple blotch, purple spot, and purple stain.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Deuteromycota, **Class:** Hyphomycetes, **Order:** Moniliales, **Family:** Dermateaceae

Reason for Inclusion in Manual

Pest Description
According to Mulder and Holliday (1975), *Cercospora kikuchii* develops a stroma, which is a compact mass of somatic hyphae, made up of few to many irregular to round, brown cells. The stroma develops within the seed coat on germinating seeds. On leaves, the stroma develops just below the epidermis. On stems and petioles, the stroma is deep within the cortex. Fascicles of conidiophores arise directly from the stroma in clusters of 2 to 5 or more. Conidiophores are divergent, medium brown in color but paler towards the apex, unbranched, geniculate, prominent, conidial scars present, septate, and 45 to 220 x 4 to 6 µm in size (longer in culture). Conidia (sympodulospores) are hyaline, acicular with 0 to 22 septa, truncate base, can be straight or curved, apex tapered, thickened hilum, and 50 to 375 x 2.5 to 5 µm in size. In culture, young hyphae are hyaline, septate, 2 to 4 µm wide, granular and sometimes noduled, whereas, older

hyphae are pale brown, 3 to 5 µm wide and closely septate. In older cultures, orange-brown colored thick-walled cells resembling chlamydospores (6 to 15 µm diam.) develop (Schuh, 1999). *C. kikuchii* has several vegetative compatibility groups (Cai and Schneider, 2005).

Biology and Ecology

The source of the primary inoculum of *C. kikuchii* is the infected crop residue of the previous year. The pathogen sporulates readily on overwintered soybean stems on the soil surface. Conidia on fallen leaves from early infections provide the secondary inoculum. Weeds are also a potential source of inoculum (Jones, 1959; Roy, 1982). In water, conidia germinate within 2 to 3 hours with one or more germ tubes. Germ tubes enter leaf tissue through stomata or penetrate the epidermal cells directly. These infections are mostly latent and only a few cells are colonized. Desiccation of leaves and the subsequent remoistening cause a rapid growth and sporulation of the fungus. Infections resulting from stomatal penetration are not visible until growth stage R4 (Schuh, 1999).

C. kikuchii enters the seeds when pod maturation begins. The hilar region is a major site for direct penetration, but it may also occur through pores in the seed coat. In naturally-infected seeds, the fungus resides primarily in layers of the seed coat and rarely is found in the embryo and cotyledons (Ilyas et al., 1975).

The occurrence of *C. kikuchii* in seeds is influenced by temperature, duration of pod wetness period, and the developmental stage of the soybean pods. The optimum temperature for infection is 25°C, with no infection developing below 15°C or above 35°C. In general, seed infection increases with increasing pod wetness periods of up to 30 hours. No disease develops at pod wetness periods of <24 hours (Schuh, 1992). Seed lots can contain 100% of the soybean seeds with symptoms of purple stain. Many infected seeds do not show symptoms. Stained seeds germinate poorly (Yeh and Sinclair, 1982).

Figure 1. Symptoms of purple seed stain. Photo Courtesy of X. B. Yang.

Measures to control *C. kikuchii* include: planting of less susceptible cultivars (Schuh, 1999), using fungicides to control both seed and leaf infection, and treating seeds with hot vegetable oil (90°C for 5 min) or hot water (49°C for 5 min) (Pyndji et al., 1987).

Pest Importance

Purple seed stain or Cercospora leaf blight occurs worldwide, wherever soybeans are grown. The seed phase of the disease does not adversely affect

yields. Discoloration of seeds does affect marketability, whether it is used for planting or processing. Substantial crop losses have been attributed to the leaf phase of the disease in southern regions of the U.S. (Walters, 1980). Losses of 3.5 and 0.5 million metric tons were estimated for 1978 and 1979, respectively, for the 15 southern states in the U.S. (Schuh, 1999).

Symptoms/Signs

The most characteristic symptom of the disease is the light to dark purple discoloration of the seed coat (Fig. 1). This discoloration may vary from tiny spots to spots covering the whole seed coat. Wide cracks in the seed coat, usually extending transversely along the seed, are often present. When an infected seed germinates, cotyledons become shriveled and dark purple within 10 to 15 days of planting. Infections on the cotyledons spread to stems of young seedlings, where the infection produces a dark diseased area, which may later encircle the stem and kill the seedling. Surviving plants are often stunted and appear weaker than healthy plants.

Figure 2. Symptoms of Cercospora leaf blight. Photo Courtesy of X. B. Yang.

In primary and secondary leaves, angular to irregular reddish-purple spots (Fig. 2) occur on both upper and lower leaf surfaces. Spots may vary from a pin-point in size to irregular patches up to 1 cm in diameter. When infections are numerous, leaves become prematurely yellow. Upper leaves exposed to the sun may initially show a light purple appearance. Later, this discoloration deepens and extends over the entire upper leaf surface, giving the affected leaves a leathery, dark, reddish-purple appearance.

On older plants, slightly sunken, irregular, reddish-purple areas (1 to several mm long) may appear on stems and petioles. Infected areas may coalesce to completely encircle the stem or petiole, and premature defoliation may occur. Maturing pods show minute, reddish to reddish-purple areas, which later become purplish-black.

Known Hosts

Major hosts

Glycine max (soybean).

Minor hosts

Cyamopsis tetragonoloba (Cluster bean or guar), *Jacquemontia tamnifolia* (small flower morning glory), *Phaseolus* spp. (beans), *Senna obtusifolia* (sicklepod), *Vigna spp.* (cowpea), and *Xanthium strumarium* (common cocklebur).

Known Distribution

CABI International (2004) list of countries where purple seed stain or Cercospora leaf blight has been found: **Asia:** Bangladesh, China, India, Iran, Japan, Korea, Malaysia, Nepal, Pakistan, Sri Lanka, and Thailand. **Europe:** France and USSR. **Africa:** Cameroon, Ethiopia, Gabon, Ghana, Liberia, Mozambique, Nigeria, South Africa, Togo, Uganda, Zambia, and Zimbabwe. **North America:** Canada and the U.S. **Caribbean:** Cuba, Jamaica, Puerto Rico, and Trinidad and Tobago. **South America:** Argentina, Bolivia, Brazil, and Colombia. **Oceania:** Australia, Fiji, and Papua New Guinea.

Potential Distribution Within the US

Purple seed stain was first reported in the U.S. in Indiana in 1924. At present, the disease is either localized or widespread in Arkansas, Delaware, Florida, Illinois, Indiana, Iowa, Kentucky, Louisiana, Maryland, Michigan, Mississippi, Missouri, North Carolina, Ohio, South Dakota, Tennessee, and Virginia,

Survey

No specific survey methods for purple seed stain or Cercospora leaf blight are currently available. Any survey method for foliar diseases in soybean may be applicable. Survey would most likely be based on visual symptoms.

Pods: Minute, reddish to reddish-purple lesions.

Seeds: Distinct pink to pale or purple areas from specks to large blotches (Fig.1).

Seedling: Shriveled cotyledons, usually dark purple; seedlings fall prematurely; stems girdled; and the young plant is either killed or stunted.

Stem and petiole: Reddish-purple, slightly sunken lesions several mm. long, dieback.

Leaf: Small, red-purple, angular lesions on both sides of sun-exposed upper leaves during seed set. Leaf may have a leathery appearance highlighted with bronzing. Premature defoliation may be present.

Whole plant: Stunted or killed.

Key Diagnostics

C. kikuchii is conspicuous and easily diagnosed. On diseased cotyledons and stems, it develops a velvety, grayish-white growth of conidiophores and conidia within 1 to 2 weeks after germination. On infected leaves and seed coats, *C. kikuchii* sporulates abundantly under high humidity and temperatures of 23°C to 27°C when incubated in alternating 12-hour periods of light and darkness for 48 to 72 hours (Schuh, 1999). *C. kikuchii* can be isolated on culture media such as potato dextrose agar or V-8 juice agar. It develops colonies with white margins, which become grayish olive toward the center. Deep folds radiate from the center

of the colony. The medium under the colony is often dark purple with pink margins. Conidiophores are yellowish-brown to dark brown at the base, paler towards the apex, and hyaline at the tip; and conidia are hyaline, acicular with 0 to 22 septa (Mulder and Holliday, 1975).

To detect *C. kikuchii* in seeds, Bradley et al. (2002) surface-sterilized a sample of 40 seeds in a 0.5% solution of sodium hypochlorite for 4 min. and then rinsed twice for 5 min. in sterile distilled water. Seeds were then plated in a 9-cm petri dish containing potato dextrose agar and incubated at 25°C for 5 days. *C. kikuchii* was recognized by the intense maroon-to-purple pigment developing from the purple stain on the seed coat.

McGee and Nyvall (1984) developed the blotter method in which a sample of 400 seeds is surface sterilized in 1% sodium hypochlorite for 30 seconds and then rinsed in sterile water. Sterilized seeds are incubated on a moistened blotter at 25°C for 10 days under continuous light. Seeds are evaluated for the presence of *C. kikuchii* as indicated by the purple staining on the seed coat under fungal growth.

Differentiation from Asian soybean rust

In Cercospora leaf blight, only the upper leaf surface is discolored and no pustules are found on the underside of the leaf. Pustules of Asian soybean rust are clustered alongside the veins and have pores from which masses of uredionospores are released.

Colletotrichum lindemuthianum

Scientific Name
Anamorph: *Colletotrichum lindemuthianum* Sacc. and Magnus
Teleomorph: *Glomerella cingulata* (Stonem.) Spauld. Et Screnk

Synonyms:
Anamorph: *Gleosporium lindemuthianum, Gleosporium socium*
Teleomorph: *Glomerella cingulata* f. sp*. phaseoli, Glomerella lindemuthiana*

Common Name(s)
Anthracnose of bean, anthracnose of legumes, pod canker of bean, dry bean anthracnose, pea anthracnose

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Deuteromycota, **Class:** Coelomycetes, **Order:** Melanconiales, **Family:** Melanconiaceae

Reason for Inclusion in Manual

Pest Description
Bean anthracnose is caused by *Colletotrichum lindemuthianum*. *Glomerella cingulat*a (Phylum Ascomycota, Class Ascomycetes, Order Phyllachorales, Family Phyllachoraceae) has been reported as the teleomorph (perfect, sexual stage) of this pathogen, but it is rarely found in culture or in nature and the name of the anamorph (imperfect stage) is commonly used.

The teleomorph was first found in cultures obtained from beans with anthracnose symptoms by Shear and Wood (1913). Although pathogenicity was not demonstrated in their isolates, they believed the isolates constituted the teleomorph of *C. lindemuthianum* and named it *Glomerella lindemuthianum*. In 1970, the teleomorph was rediscovered by pairing two isolates to produce ascomata (perithecia). Because the teleomorph-producing isolates were

pathogenic only to beans and their morphology was indistinguishable from *G. cingulata*, they were named *G. cingulata* f. sp. *phaseoli* (CABI, 2004; Pastor Corrales and Tu, 1989). The sexual stage has been observed in a few instances in eastern Canada but not in the U.S. The production of a sexual stage could result in the creation of new races. These races are capable of attacking specific resistance genes incorporated in commercial cultivars.

The fungus *C. lindemuthianum* is almost exclusively a pathogen of dry edible beans but can infect other legumes, including soybean. Conidia (spores) are produced in open structures called condiomata (acervuli) that are located in the center of lesions, which may be present on pods, leaves, stems, and branches. The acervuli are round or elongated, up to 300 μm diameter. They may be intraepidermal or subepidermal, disrupting outer epidermal cell walls of the host. Occasionally, cells of the acervuli develop as setae, which are brown, septate, and slightly swollen at the base, tapering gently to a rounded paler apex. Setae are 4 to 9 μm wide and usually <100 μm long. They may be present in culture or on the host at the margin of the condiomata. Condiomata have pale salmon-colored spore masses. Conidia are unicellular, hyaline, cylindrical with both ends obtuse or with a narrow and truncate base. Condia are uninucleate and usually have a clear vacuole-like body near the center. Reported conidial measurements are in a range of 11 to 22 x 2.5 to 5.5 μm. Conidia are formed on unbranched unicellular hyaline or pale brown cylindrical conidiophores (40 to 60 μm).

The pathogen has been proposed as a model organism for the analysis of plant-pathogen interactions, because it is hemibiotrophic and has been used to study many aspects of plant-pathogen interactions including phytoalexin production, cell-wall degrading enzymes, and infection processes (Perfect et al., 1999).

Biology and Ecology

Infection by *C. lindemuthianum* is favored by moderate temperatures between 13 and 26°C (Lauritzen, 1919; and Hwang et al., 1968;), with an optimum of 17 to 24°C (Tu and Aylesworth, 1980). Infection and development of the pathogen is delayed or prevented by temperatures outside the range of 7 to 33°C (Salazar and Andersen, 1969; Rahe and Kuc, 1970; Tu and Aylesworth, 1980). Humidity of more than 92% or free moisture is required during all stages of conidium germination, incubation and subsequent sporulation.

A conidium germinates in 6 to 9 hours under favorable environmental conditions to form a germ tube and appressorium, which attaches to the host cuticle by a gelatinous layer. The pathogen penetrates the cuticle and epidermis mechanically. Following penetration of host cells, when temperatures are favorable, infectious hyphae enlarge and grow between the cell wall and protoplast for 2 to 4 days without apparent damage to host cells.

Several days later, cell walls are degraded, probably by L-galactosidase (English and Albersheim, 1969) and protoplasts disorganize and collapse. Water-soaked

lesions appear (Mercer et al., 1975) which later turn dark brown because of high tannin content. The mycelium may then mass within the lesion site and form acervuli, which rupture the host cuticle. The acervuli contain a stromatic layer of three to 50 conidiophores, depending upon the lesion size. Numerous conidia are formed and embedded in a water-soluble gelatinous matrix. Newly produced conidia are more infectious than older ones.

C. lindemuthianum can overwinter either in seed or infected crop residues. C. *lindemuthianum* survives as dormant mycelium within the seed coat, sometimes even within cells of cotyledons, as spores between cotyledons or elsewhere in the seed (Zaumeyer and Meiners, 1975). It can survive for at least 2 years in seed (Mordue, 1971a, 1971b). Depending on environmental conditions, longevity in infected pods and seeds varies considerably. Moisture is an important factor that influences the survival of the fungus. The pathogen was able to survive at least 5 years on pods and seeds that were air-dried and kept in storage at 4°C or on dry, infected plant materials left in the field in sealed polyethylene envelopes that prevented contact with water (Tu, 1983). An alternating wet/dry cycle was detrimental to fungal survival. Since the pathogen can be seed-borne, epidemics can start at any stage of development of the crop.

Differential susceptibility of bean cultivars to various isolates of the pathogen was first reported by Barrus (1911). Isolates were classified on the basis of host reaction when inoculated with isolates of *C. lindemuthanium* as two distinct races: alpha and beta. Since that time, surveys have confirmed the extensive variation in the pathogen. Race designation is based on the reaction of different host cultivars when inoculated with isolates of the pathogen. Lack of standardization of the cultivars used in this determination has made comparison of race designation difficult (Zaumeyer and Meiners, 1975). Races identified in North America include alpha, beta, gamma, epsilon and lambda (Burkholder, 1923, Leach, 1923, Andrus and Wade, 1942; and Tu et al., 1988). Recently a new race, alpha-Brazil, has been recognized in Canada and the U.S. (Tu, 1994; Kelly et al., 1994). Other races have been reported from Europe, Latin America, and Africa.

Initial infection comes from the fungus present in the seed or dry crop debris. The fungus survives in the seed as long as the seed remains viable. After initial infection, moisture, in the form of rain, dissolves the water-soluble gelatinous matrix in which the conidia rest in the acervulus. Moderate rainfall at frequent intervals, particularly when accompanied by wind or splashing rain, is essential for local dissemination of conidia and for development of severe anthracnose epidemics. In Ontario, Canada, Tu (1981) found that *C. lindemuthianum* required about 10 mm of rain to establish infection. Long-distance dissemination (3 to 5 meters) may result from splashing raindrops blown by gusting winds. Conidia also may be dispersed within the crop by movement of insects, animals and man, especially when foliage is moist.

Pest Importance

Anthracnose of dry bean is a potentially devastating seed-borne disease that affects dry beans in temperate regions of the world. The disease can affect any above-ground part at any stage of development. However, early infections usually result in heavier yield losses and higher seed transmission rates. Weather conditions favorable to anthracnose development can result in complete loss of the crop.

Anthracnose is an important fungal pathogen of *P. vulgaris,* affecting yield, seed quality, and marketability of the crop. The disease causes greater losses in temperate and subtropical zones than in the tropics. It has caused economic losses in North, Central and South America, Europe, Africa, Australia and Asia. It is one of the major constraints to production in regions where the crop provides an essential part of the people's daily diet, especially in Latin America and Africa. Yield losses of 95% have been recorded in Columbia and over 92% in Malawi (Allen, 1983). At one time, it was considered the most important disease in the bean-producing areas of eastern U.S.; losses amounting to $1.5 million were reported in Michigan during 1914. The disease declined considerably in importance during 1955 to 1976, due to the widespread use of clean seed produced in areas where anthracnose does not occur. However, during 1977 to 1978, a serious epidemic occurred in southwestern Ontario, Canada, due to the introduction of the gamma and delta races of the pathogen (Tu, 1988). Clean seed and resistant cultivars have also diminished the importance of bean anthracnose in western Europe.

Symptoms/Signs

Bean: Symptoms of anthracnose can appear on any plant part. In the early stages of disease development, lesions usually appear on the lower leaf surface along the veins, which show brick red to purplish red to dark brown discoloration (Fig. 1). In later stages of disease development, lesions can be observed on both sides on the leaf.

Figure 1. Lesions on lower leaf surface along the veins. Photo courtesy of C. Bradley, North Dakota State University.

Dark brown eyespots that develop longitudinally along the stems are an early sign of stem infection. In the young seedlings, the eyespots enlarge and the stem may break off. On older stems, the eye-shaped lesion is limited to the approximate length of 5 to 7 mm. and the lesion often has a sunken cankerous center.

When the fungus infects pods (Fig. 2A), it usually produces small round lesions (cankers), about 1/8 inch in diameter, with a sunken center (Fig. 2A). Sporulation can occur in lesions on the petiole and larger leaf veins, thereby producing

secondary inoculum. In wet weather, masses of spores, salmon-tan to orange in color may cover the center of the cankers. The borders of the cankers are usually well defined and dark brown in color. When the fungus attacks the pods in their early stages of development, it can penetrate and infect the newly forming seeds. If severely infected, young pods shrivel and dry up. Infected seeds may be shriveled, discolored, and have dark brown to black cankers (Fig. 2B). Discolored areas are more conspicuous on white-coated seeds.

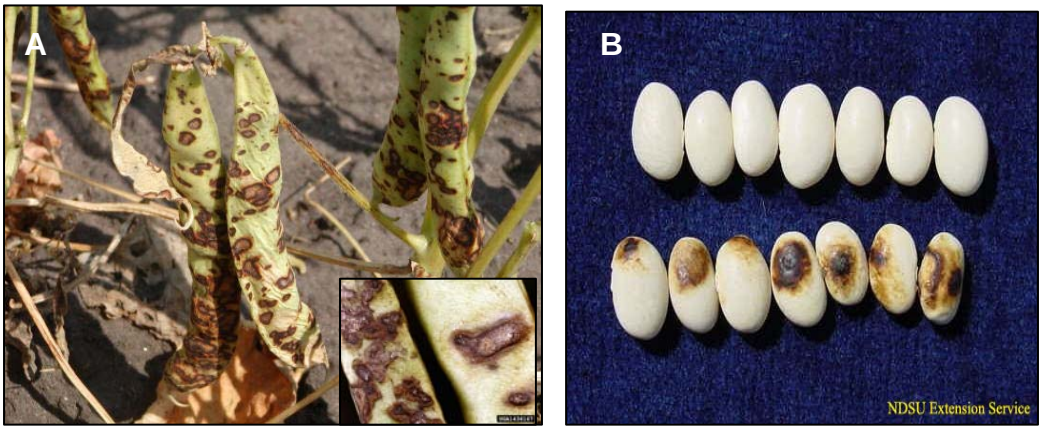


Figure 2. *C. lindemuthianum* symptoms on pods and seeds. Photos courtesy of C. Bradley, North Dakota State University and 1707Hwww.invasive.org.

Infections occurring late in the season may result in infected seeds that do not show symptoms. However, seedlings produced by symptomless seeds could become sources of infection for new plantings. Infected seeds harbor the fungus under the seed coat, where chemical seed treatments may not come in contact with the fungus to provide an adequate level of control.

Soybean: *C. lindemuthianum* causes dark brown elongate more or less angular spots along the veins on the petioles, stem and lamina. When infection occurs on the hypocotyls, the plant collapses. Seeds when infected turn brown or black.

Known Hosts

Major hosts

Cajanus cajan (pigeon pea), *Phaseolus* spp. (beans), and *Vigna sinensis* ssp. *sesquipedialis* (asparagus bean).

Minor hosts

Glycine max (soybean), *Lablab purpureus* (hyacinth bean), *Lens culinaris ssp. culinaris* (lentil), *Pisum sativum* (pea), *Vicia faba* (broad bean), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), and *Vigna unguiculata* (cowpea).

Known Distribution

The disease has a worldwide distribution and occurs wherever *P. vulgaris* is grown. Countries known to have *C. lindemuthianum* include: **Asia:** Armenia,

Azerbaijan, Bangladesh, Brunei Darussalam, China, India, Indonesia, Iran, Japan, Korea, Malaysia, Myanmar, Nepal, Oman, Pakistan, Philippines, Saudi Arabia, Thailand, Turkey, Vietnam, **Europe:** Austria, Bulgaria, Czech Republic, Denmark, Finland, France, Germany, Hungary, Poland, Romania, Russian Federation, **Africa:** Burundi, Congo Democratic Republic, Ethiopia, Kenya, Libya, Madagascar, Malawi, Mali, Mauritius, Morocco, Mozambique, Nigeria, Rwanda, Senegal, South Africa, Tanzania, Uganda, Zambia, Zimbabwe, **North America:** Canada, Mexico, the U.S., **Central America:** Costa Rica, Guatemala, Honduras, Nicaragua, Argentina, Brazil, Columbia, Ecuador, Peru, **Oceania:** Australia, New Caledonia, New Zealand, and Papua New Guinea.

Potential Distribution Within the US

Anthracnose on dry bean occurs on bean in Alaska, Alabama, California, Delaware, Florida, Georgia, Hawaii, Idaho, Illinois, Iowa, Kansas, Michigan, Mississippi, New York, North Carolina, North Dakota, Oklahoma, Oregon, South Dakota, and Texas. There is very limited information on the distribution of dry bean anthracnose on soybean within the U.S.

Survey

The disease is detected by visually inspecting the underside of leaves for small, angular, reddish to purplish-brown lesions developing predominately along veins and pods for sunken lesions. Samples of leaves and pods showing characteristic symptoms and with visible setae (Fig. 3) are commonly plated on acidified potato dextrose agar and colony morphology is examined (Gonzalez et al.,1998)

Figure 3. Setae of *Colletotrichum* spp. Photo courtesy of University of Arkansas , 1708Hhttp://www.uaex.edu/Other_Areas/publications/HTML/MP197/chapter11_stem_root_diseases.asp.

C. lindemuthianum is best detected in seeds using a modified blotter test as described in ISTA Working Sheet No. 46. Surface disinfested seed in replicates of 50 seeds are placed on two layers of moistened blotter paper sheet (440 x 340 mm) and covered with a third. The sheets are then rolled and placed in plastic bags and incubated 7 days at 20 °C in darkness. After incubation, seed coats are removed and the cotyledons examined for black depressed lesions indicative of *C. lindemuthianum*. Each spot is checked for the presence of acervuli containing setae and spores.

Key Diagnostics

C. truncata, the cause of anthracnose in soybean, has been isolated from bean plants showing symptoms similar to those caused by *C. lindemuthianum*.

Seedlings of *P. vulgaris*, when inoculated with isolates of *C. truncata*, showed typical anthracnose symptoms. Conidia of this fungus were hyaline, curved and unicellular, measuring 27 x 3.5 μm; setae were also observed among the conidiophores.

Another fungus similar to *C. truncata* was isolated from bean leaves in Columbia by Pastor Corrales and Tu (1989). The leaves showed long streaks of intense reddening on the veins, but had none of the typical sunken lesions characteristic of bean anthracnose. The frequency and importance of this species has not yet been determined.

Colletotrichum truncatum

Scientific Name
Colletotrichum truncatum Schwein

Synonyms:
Colletotrichum dematium forma truncatum, Vermicularia truncate

Common names
Brown blotch of cowpea, lentil anthracnose, pod blight of soybean, soybean anthracnose, stem anthracnose of lima bean and pigeon pea

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Deuteromycota, **Class:** Coelomycetes, **Order:** Melanconiales, **Family:** Melanconiaceae

Reason for Inclusion in Manual

Pest Description
Colletotrichum truncatum is characterized by crowded, black acervular conidiomata that are borne on well-developed stromata. Conidiomata are oval to elongate, hemispherical to truncate conical, and erumpent. Setae are 60 to 300 x 3 to 8 µm, intermixed long and short, numerous, black and subulate. Conidia are borne singly on conidiophores, bluntly tapered, curved, unicellular, hyaline and 17 to 31 x 3 to 4.5 µm. Conidia usually produce one or two short germ tubes, which produce dark, sticky appressoria when in contact with the surface of the host plant or a solid surface. A narrow infection peg extends from the underside of the appressorium and penetrates the cuticle and cell wall.

Biology and Ecology:
C. truncatum is transmitted by infected soybean seeds. The pathogen was recovered from anthracnose lesions on seedlings grown in sterilized soil from naturally infected seeds (Khare and Chacko, 1983) and from inoculated seeds grown in sterilized sand under greenhouse conditions (Dhingra et al., 1978; Roy, 1982). In another report, symptomless seedlings developed in unsterilized soil in the greenhouse from seeds inoculated with a conidial suspension of *C.*

truncatum. The pathogen was later detected in the cotyledons and cortex of the stem. It then advanced longitudinally and was found in the pod and in cotyledons of developing seeds (Tiffany, 1951).

C. truncatum also survives on soybean stem residues (Lehman and Wolf, 1926). Fruiting bodies of *Colletotrichum* and a *Glomerella* teleomorph were found on 22% of 5000 stubble samples in Illinois fields (Hartman et al., 1986). Inoculum applied to soil in pots in late autumn, provided active inoculum in the following spring (Ling, 1940). Soybean cultivars A. K. (Kansas), Boone, and Williams 82 were grown in sand infested with sclerotia-forming isolates of *C. truncatum* in soil tanks at 20, 25, 30 or 35°C and at greenhouse ambient temperature (21 to 28°C). Root and hypocotyl infection were recorded on all cultivars at all temperatures. Lesion size and number generally increased with an increase in soil temperature up to 30°C and then declined. Williams 82 had the highest disease rating, Boone the lowest and A. K. (Kansas) was intermediate over all temperatures (Khan and Sinclair, 1991). Isolates of *C. truncatum* from soybean seeds produced microsclerotia in culture and in soybean tissues (Khan and Sinclair, 1992).

Inoculation studies in the field and greenhouse demonstrated transmission of the pathogen from mother plant to seed (Tiffany, 1951; Machado and Carvalho, 1975). Seed infection with *C. truncatum* was low in areas with low rainfall and high when crop populations were high (Khare and Chacko, 1983). In Puerto Rico, soybean seeds produced in the wet season were more heavily infected than those produced in the dry season (Sinclair, 1988). The role of light, temperature and relative humidity on the germination of *C. truncatum* and soybean pod infection was studied under laboratory conditions in India (Kaushal et al., 1998). The optimum temperature for conidial germination and germ tube elongation was 20°C and for soybean pod infection was 25°C. Three hours of light followed by 9 hours of dark was best for spore germination and germ tube elongation. Twelve hours of light followed by 12 hours of dark was most suitable for pod infection; pod infection and the development of acervuli took longer in continuous light. High (100%) relative humidity (RH) was required for pod infection and the development of acervuli. However, in slide germination tests at 100% RH, none of the spores germinated (Kaushal et al., 1998).

In India, the incidence of leaf infection was greater in early plantings than in late plantings (Khare and Chacko, 1983). Seedling emergence was reduced and plant infection increased when seed harvested from early plantings was used. The presence of weeds either had no effect or increased infection depending on planting time or row spacing (Hepperly, 1984). The addition of calcium to soils reduced disease development in seedlings inoculated with conidial suspensions of the pathogen under greenhouse conditions (Muchovej et al., 1980). Herbicide sprays, applied as desiccants, induced the formation of conidiomata of *Colletotrichum*, thus revealing asymptomatic infections (Cerkauskas et al., 1983).

Isolates of *Colletotrichum* and *Glomerella* species from soybeans vary in their

cultural characteristics and pathogenicity (Manandhar et al., 1984, 1988; Tiffany and Gilman, 1954). In the U.S., falcate-spored isolates of *Colletotrichum* isolated from different parts of soybean plants were separated into two colony types: *C. dematium* f.sp. *truncatum* (*C. truncatum*) and *C. capsici*. *C. capsici* was non-pathogenic or weakly pathogenic on soybean seedlings, with pathogenicity limited primarily to the cotyledons. In contrast, *C. truncatum* was extremely virulent on soybean seedlings and caused considerable pre- and post-emergence seedling death in the U.S. (Roy, 1996). Furgal-Wegrzycka (1997) studied the genetic relatedness and diversity in terms of vegetative compatability of seven species and subspecies of *Colletotrichum,* including *C. truncatum*.

C. truncatum infects other legumes, but is not highly seedborne. Microsclerotia can be disseminated several hundred meters in the dust from combine harvesters (Morrall, 1997). Soybean seed infection levels as high as 81% have been recorded in tropical and subtropical regions (Verma and Upadhyay, 1973; Fulco et al., 1979; Hepperly et al., 1983). In Florida, the pathogen was detected in 30% of 73 seed samples, with infection levels ranging from 5 to 20% (Franca Neto and West, 1989). In temperate regions such as the northern U.S., seed infection is rare despite extensive pod infection (Athow, 1987; McLean and Roy, 1988). An Illinois report indicated that less than 1% of seed was infected. The pathogen survived for over 10 years when stored at 5°C (Siddiqui et al., 1983). Histological studies have shown mycelia of the fungus in the seed coat layers, epidermis (palisade), hypodermis (hourglass) and endodermis (parenchyma) (Nik and Lim, 1984; Kunwar et al., 1985). The pathogen has been detected in wounds in the seed coat. Inoculation studies in the field and greenhouse demonstrated transmission of the pathogen from mother plant to seed (Tiffany, 1951; Machado and Carvalho, 1975).

Pest Importance

Anthracnose was first reported on soybeans in Korea in 1917. The disease is widespread in temperate soybean production zones but causes minor losses (Athow, 1987; Tiffany, 1951). Losses are more severe in warm, humid regions. In Alabama, yields were reduced by an average of 19.4% for three cultivars compared to plots in which the disease was controlled by fungicide applications (Backman et al., 1982). Yield losses of 30% were attributed to anthracnose in Nigeria in 1975 (Rheenen, 1975) and a survey in two states in Brazil detected the disease in 57% of the fields (Lehman et al., 1976). Anthracnose of maturing plants causes serious losses, particularly during the rainy period when shaded lower branches and leaves are killed.

In 1992 and 1993, *C. truncatum* was isolated from infected stems of lentil collected in North Dakota. Cultivars Crimson red and Brewer Chilean were most common in the area and both were susceptible to anthracnose. In nine of 15 fields inspected for anthracnose, severity was light to moderate. In four fields the disease was judged very severe and in two fields it was undetected (Venette et

al., 1994).

Symptoms/Signs

Soybean plants are susceptible to anthracnose at all growth stages. During early reproductive stages, irregularly-shaped, brown areas may appear on stems, pods and petioles (Fig. 1). Stems, pods and leaves may also be infected without showing symptoms. In the advanced stages of anthracnose in the late reproductive stages, infected tissues are covered with black fruiting bodies (conidiomata, acervuli) (Figs. 1, 2), which produce minute black spines (setae) that can be seen with the unaided eye.

Figure 1. Brown areas of soybean pods (left) and black fruiting bodies (acervuli) on soybean stem (right). Photo courtesy of Clemson University - USDA Cooperative Extension Slide Series. 1706Hwww.invasive.org

When pods or pedicels are infected at an early stage, seeds either do not form (pod blanking) or, if they do develop, are smaller and fewer in number.

Foliar symptoms, which develop after prolonged periods of high humidity, include necrosis of laminar veins, leaf rolling and petiole cankers. Premature defoliation may occur throughout the canopy when cankers girdle the leaf petiole. The cankers often occur where the leaflets join the petiole, resulting in a shepherd's crook. Diseased plants may be shorter than healthy plants. Affected plants senesce earlier than healthy plants, due to the combined effects of the stem and petiole lesions.

Figure 2. Acervuli on pods, arranged in concecentric rings. Photo courtesy of Ved Prakash Gupta (CABI, 2004).

Pre- and post-emergence damping-off may occur when infected seeds are planted. Dark brown, sunken cankers (lesions)

often develop on the cotyledons of emerging seedlings. These cankers gradually extend up toward the epicotyl and down to the radicle. During humid weather one or both cotyledons may become water-soaked, wither and fall off. Infection may spread from the cotyledons to the young stems where small, deep-seated cankers form, often killing the seedling.

Seeds colonized by *C. truncatum* may not show any symptoms but can develop brown staining or small, irregular, grey areas with black specks. The fungus is confined at first to the seed coat. The infected seeds may die during germination or, if they germinate, may produce infected seedlings.

Known Hosts

Major hosts

Aeschynomene americana (American jointvetch), *Arachis hypogaea* (peanut), *Bryophyllum pinnatum* (air plant), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Centrosema pubescens* (centro), *Clitoria ternatea* (butterfly-pea), *Crotalaria juncea* (sunn hemp), *Desmodium* (tick clovers), *Glycine max* (soybean), *Lens culinaris* ssp*. culinaris* (lentil), *Medicago sativa* (alfalfa), *Panax ginseng* (Asiatic ginseng), *Phaseolus* (beans), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Stylosanthes guyanensis*, *Trifolium pratense* (purple clover), *Trifolium subterraneum* (subterranean clover), *Vicia sativa* (common vetch), *Vigna* (cowpea), *Vigna aconitifolia* (moth beans), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), *Vigna unguiculata* (cowpea), and *Zornia diphylla* (trencilla).

Minor hosts

Abutilon theophrasti (velvet leaf), *Amaranthus hybridus* (smooth pigweed), *Apocynum cannabinum* (hemp dogbane), *Asclepias* (silkweed), *Chenopodium album* (fat hen), *Datura stramonium* (jimsonweed), *Ipomoea* spp. (morning glory), *Polygonum* (knotweed), *Solanum* (nightshade), and *Xanthium strumarium* (common cocklebur)

Known Distribution

This disease is present throughout most soybean production areas worldwide including: **Asia:** Bangladesh, China, Republic of Georgia, India, Indonesia, Iran, Japan, Korea, Malysia, Nepal, Pakistan, Philippines, Saudi Arabia, Singapore, Thailand, Turkey, United Arab Emerates, Vietnam, **Europe:** Bulgaria, Former Yugoslavia, France, Hungary, Italy, Moldova, Russian Federation, Serbia and Montenegro, Slovakia, Spain, **Africa:** Burkina Faso, Cameroon, Egypt, Ethiopia, Gabon, Ghana, Guinea, Ivory Coast, Kenya, Madagascar, Malawi, Mauritius, Mozambique, Nigeria, Senegal, Sierra Leone, South Africa, Tanzania, Uganda, Zambia, Zimbabwe, **North America:** Canada, the U.S., **Central America:** Barbados, Belize, Cuba, Guatemala, Honduras, Puerto Rico, Trinidad and Tobago, **South America:** Argentina, Brazil, Columbia, Guyana, Venezuela, **Oceania:** Australia, Federated States of Micronesia, Fiji, Papua New Guinea, Samoa, and Tonga (CABI, 2004).

Potential Distribution Within the US

The disease is currently present in Alabama, Arkansas, Florida, Georgia, Hawaii, Illinois, Indiana, Kansas, Kentucky, Louisiana, Maryland, Minnesota, Mississippi, Missouri, North Carolina, North Dakota, Oklahoma, Pennsylvania, South Dakota, Tennessee, Texas, Washington and Wisconsin.

Survey

Survey is conducted primarily by a visual survey of plant symptoms.

Seedling: Pre- and post-emergence damping-off. Dark-brown, sunken lesions often form on cotyledons and may extend upward towards the epicotyl or downward to the radicle.

Stem petiole and pod: Irregular blotches that are covered with black acervuli at later growth stages.

Leaf: Leaf rolling, necrosis of laminar veins, and premature defoliation.

Seed: Shriveled or moldy, with brown discoloration or small, irregular grey areas with black specks

Key Diagnostics

Early soybean infection may result in pods without seeds, or fewer, smaller seeds. Look for pod cavities which may be completely filled with mycelium of the anthracnose fungus. Seeds may appear moldy, dark brown and shriveled.

Anthracnose of soybean can be easily identified by the presence of black fruiting bodies (acervuli) on infected tissues. The acervuli produce minute spines (setae), which are diagnostic for preliminary identification of the pathogen.

Culture *C. truncatum* on oatmeal agar or potato-dextrose agar under alternating 12-hour periods of light and dark. Optimum temperature for growth is 25°C. Whitish colonies are produced which eventually turn smoky black with abundant conidiomata (CABI, 2004).

Disported phaseolorum var. *sojae*

Scientific Name
Teleomorph: *Diaporthe phaseolorum* var. *sojae* Lehman Wehm.
Anamorph: *Phomopsis phaseoli* var. *sojae* Lehman Sacc

Synonyms:
Teleomorph: *Diaporthe sojae*
Anamorph: *Phomopsis sojae*

Common Name(s)
Pod and stem blight.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota, **Class:** Ascomycetes, **Order:** Diaporthales, **Family:** Valsaceae

Reason for Inclusion in Manual

Pest Description
The pycnidial conidiomata of *D. phaseolorum* var. *sojae* is stromatic, black, solitary or aggregated, and usually unilocular. Pycnidia are mutic or have beaks <200 µm long with apical ostioles. Locules are uni to multi-ostiolate, lenticular, and up to 300 µm wide (Pioli et al., 2003). Alpha conidia are 1.5 to 3.5 x 5.5 to 10.5 µm in size, hyaline, usually fusiform and biguttulate. Beta conidia are hyaline, filiform and hamate. Alpha conidia are the main source of asexual inoculum, whereas beta conidia are infrequent. Perithecia are produced on over-seasoned soybean stems. Mature perithecia are usually solitary, spherical and slightly flattened at the base, with long, tapered beaks. Asci are elongate, clavate, and dissolve before releasing ascospores. The 2 to 6 x 9 to 13 mm in size ascospores are fusiform, elliptical, bicellular and biguttulate (Kulik and Sinclair, 1999). Ascospores and alpha conidia germinate readily and produce white cottony mycelium. Single spore cultures (homothallic) eventually produce perithecia on agar (Jensen, 1983). *D. phaseolorum* var. *sojae* develops floccose, white to whitish-grey colonies with yellowish tonalities and rope-like mycelia on

acidified potato dextrose agar (Kulik and Sinclair, 1999). The underside is grey or tan to dark brown with black pulvinate stromata scattered in the medium (Pioli et al., 2003). The optimal colony growth is between 25°C to 28°C.

Biology and Ecology

D. phaseolorum var. *sojae* overwinters on infected soybean residues (stems, pods and/or seeds), which serves as the primary inoculum to infect seedlings (Kmetz et al., 1979). The infected seeds usually do not germinate, but if they germinate, seedlings are weak (Athow and Laviolette, 1973). In most infected seeds, if the radicle emerges, it is rapidly killed by the pathogen (Ellis et al., 1974). Pycnidia with alpha conidia are abundant on overwintered soybean residues, as well as on current season plant debris. Perithecia are produced in early summer on overwintered stem debris (Kulik and Sinclair, 1999). The pathogen first infects the petioles of the lower leaves and broken lower branches. Alpha conidia can be found on the surface of immature, symptomless soybean plants. Alpha conidia are primarily found on the lower third of the plants, but may be found in plants growing up to 2 m from the source of inoculum (Kmets et al., 1979). Maximum spore production occurs during pod filling. The appearance of stem blotching, pod blotching, and pycnidia coincides with premature ripening of most plants in the field.

The stage of pod senescence is critical for seed colonization by the pathogen. The pathogen does not appear to become systemic from infected seeds and can remain passive, without being detected, before causing slightly premature ripening and/or the appearance of pycnidia. *D. phaseolorum* var. *sojae* enters pods through abrasions, cracks or other injuries. Seed infection is higher on lodged or broken branches in contact with soil. Seed infection also increases with the delay of harvest and alternating wetting and drying conditions that are conducive for pod deterioration and splitting along the suture (Athow and Laviolette, 1973). In the seed coat, the mycelium of the pathogen is abundant in the hourglass cell layer and less so in the parenchyma and palisade cell layer. The fungus is occasionally found in the cotyledonary tissues (Ilyas et al., 1975).

Pod and stem blight can be controlled by planting resistant lines to *D. phaseolorum* var. *sojae*, practicing clean tillage, removing host debris, harvesting on time, and rotating soybean with non-host crops. Although little control of the disease can be achieved by planting Phomopsis-free seeds in endemic areas, it may be of practical importance in preventing the introduction of the pathogen to new areas. Seed treatment with fungicides may reduce the incidence of the pathogen and increase germination in both symptomatic and asymptomatic seeds. Resistant lines to *D. phaseolorum* var. *sojae* have a common lineage to PI227687 and PI229358 and the Brazilian cultivar Santa Maria.

Pest Importance

D. phaseolorum var. *sojae* is generally considered a weak parasite. The most important aspect of pod and stem blight is its effect on seed quality. Before 1960,

D. phaseolorum var. *sojae* was considered of little importance to soybean production. Since then, significant germination losses have been reported in Canada, Brazil and the U.S. (Ellis et al., 1974; Wallen and Seaman, 1963). If harvest is delayed, seed infection on susceptible cultivars can exceed 50% (Wilcox et al., 1974). Oil from infected seeds has a rancid, off-odor smell, and a high peroxide value indicating oil deterioration (Hepperly and Sinclair, 1978).

Symptoms/Signs

D. phaseolorum var. *sojae* infects stems, petioles, pods, seed, and less frequently leaves. Symptoms include poorly developed pods, broken branches, petioles and leaves (Athow and Laviolette, 1973). Lesions on cotyledons of seedlings from infected seeds are almost colorless to bright red or brown (Kulik and Sinclair, 1999). On the hypocotyl at or below the soil line, lesions are reddish brown streaks up to 1.5 cm long. Under field conditions, no definite leaf or stem lesions are produced. Plants nearing maturity develop large numbers of black specks and fungal fruiting bodies (pycnidia) in straight rows along the main stem (Fig.1). Pods with pycnidia always contain infected seeds, which are smaller and may have brown or black spots on the seed coat. They also exhibit varying degrees of cracking on the seed coat, wrinkling, discoloring, flattening, and are frequently covered with white mold (Athow and Laviolette, 1973; Ellis et al., 1974).

Figure 1. Symptoms of pod and stem blight. Photo Courtesy of Clemson University-USDA Cooperarive Extension Slide Series. 1709Hwww.invasive.org

Known Hosts

Major hosts

Glycine max (soybean)

Minor hosts

Abelmoschus esculentus (okra), *Allium cepa* (onion), *Allium sativum* (garlic), *Arachis hypogaea* (peanut), *Capsicum frutescens* (chili), *Lespedeza* spp., *Lupinus* spp., *Lycopersicum esculentum* (tomato), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean) and *Vigna unguiculata* (cowpea).

Wild hosts

Abutilon theophrasti (velvet leaf) and *Amaranthus spinosus* (spiny amaranth).

Known Distribution

CABI International (2004) list of countries where pod and stem blight is reported: **Asia:** Armenia, Azerbaijan, China, Georgia, India, Israel, Korea, Malaysia, Nepal, Sri Lanka, Taiwan, and Thailand. **Europe:** Bulgaria, Czech Republic, Yugoslavia, France, Hungary, Italy, Moldova, Romania, Serbia and Montenegro, Spain, and the USSR. **Africa:** Cameroon, Egypt, Malawi, Nigeria, Senegal, Sierra Leone, South Africa, and Tanzania. **North America:** Canada and the U.S. **Central America:** Cuba. **South America:** Brazil, Colombia, Ecuador, Guyana, Paraguay, and Venezuela. **Oceania:** Australia.

Potential Distribution Within the US

The pathogen is present in Alabama, Arkansas, Delaware, Florida, Georgia, Hawaii, Illinois, Indiana, Louisiana, Maryland, Michigan, Mississippi, Missouri, New York, North Carolina, Ohio, Oklahoma, South Carolina, Texas, and Virginia.

Survey

No specific survey methods are available for stem and pod blight. The disease is frequently recognized in the field by the premature ripening and development of pycnidia on infected stems and poorly developed pods at maturity (Athow and Laviolette 1973). Symptomatic stems have black specks and fungal fruiting bodies (pycnidia) in straight rows (Fig.1).

Key Diagnostics

Several methods are available for isolation of *D. phaseolorum* var. *sojae* from stems and pods. Pieces of these plant parts are usually washed for 2 hours under running tap water, surface-sterilized by dipping in 95% ethanol for 10 seconds and then in 0.25 to 0.5% sodium hypochlorite for 4 min., and washed twice in sterile, deionized water. Then, tissue pieces are plated on potato dextrose agar amended with streptomycin sulfate and tetracycline, and incubated for 5 days in the dark at 25±2°C (Hepperly and Sinclair, 1978; Bisht and Sinclair, 1985).

D. phaseolorum var. *sojae* grows easily on natural potato dextrose agar or synthetic media at 15°C to 32°C (optimum 28°C) and over a wide range of pH (4 to 7). Development of pycnidia is enhanced by exposing colonies to continuous light (2500 lux) or alternating 12 hour light-12 hour dark (Sar et al., 1979). Jensen (1983) found that cultures grown on PDA at 21°C with 12 hours of daily fluorescent light produced pycnidia with both alpha and beta conidia, and also perithecia and asci of *D. phaseolorum* var. *sojae*.

Identification of suspected isolates can be done by the procedure outlined by Pioli et al. (2003). Pathogenicity is tested by inserting a toothpick overgrown with mycelium of a suspected isolate into the stem at the first or second trifoliate leaf node and covering the wound with petrolatum (Athow and Laviolette, 1973; Kmetz et al., 1979).

Two molecular methods have successfully been applied to differentiate *D. phaseolorum* var. *sojae* from *P*. *longicolla*, *D. phaseolorum* var. *meridionalis*, *D. phaseolorum* var. *caulivora*, *D. phaseolorum* var. *sojae* and *Phomopsis* spp. (Zhang et al., 1997; Zhang et al., 1999). A specific band for *D. phaseolorum* var. *sojae* was observed from DNA extracts of tissue samples from symptomless plants inoculated with *P*. *longicolla* and *D. phaseolorum* var. *sojae* (Zhang et al., 1997). A species-specific detection of *D. phaseolorum* var. sojae from soybean seeds was also possible using polymerase chain reaction-restriction fragment length polymorphism (PCR-RFLP) and TaqMan chemistry (Zhang et al., 1999).

Fusarium graminearum

Scientific Name
Anamorph: *Fusarium graminearum* Schwabe
Teleomorph: *Gibberella zeae*

Synonyms:
Fusarium roseum, *Fusarium roseaum* f. sp. *cerealis*, *Gibbera saubinetti*, *Giberella saubinetti*, *Sphaeria zeae*, *Fusarium roseum* var. *graminearum*

Common Name(s)
Cobweb disease, ear rot of maize, Fusarium root and stalk rot, Gibberella ear rot, Gibberella stalk rot, head blight, headblight of maize, malformation disease, pink ear rot, red ear rot, pink mold, root rot of maize, scab of maize, stalk rot of maize, tombstone scab, and whiteheads

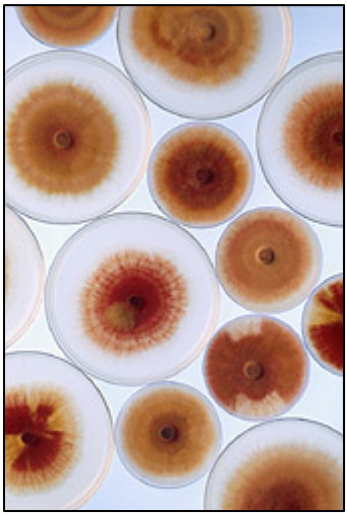


Figure 1. *Fusarium graminearum* cultures. Photo courtesy of USDA-ARS Cereal Disease Laboratory.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota, **Class:** Ascomycetes, **Order:** Hypocreales, **Family:** Nectriaceae

Reason for Inclusion in Manual


Pest Description
Gibberella zeae is the cause of Fusarium head blight or scab on cereals and of ear and stalk rot of maize. The anamorph of *G. zeae* is *Fusarium graminearum*. The cause of root and crown rot of cereals was also known as *F. graminearum* Group 1, but it is now recognized as a separate species recently re-described as *F. pseudograminearum* by Aoki and O'Donnell (199a). The teleomorph of *F. pseudograminearum* (*Gibberella coronicola*) has since been described (Aoki and O'Donnell, 199b).

Anamorph: The anamorph, *F. graminearum*, is most commonly found in nature and can be readily isolated from infected material (Fig. 1). Microconidia are absent. Macroconidia are produced from doliiform conidiogenous cells 10 to 14 x 3.5 to 4.5 µm, formed laterally or on short multibranched solitary conidiophores; sporodochial conidiomata may form in older cultures. Conidia are often formed sparsely; they are falcate, sickle-shaped or markedly dorsiventral, 3 to 7 septate, 25 to 50 x 3 to 4 µm, with a well developed, often pedicellate foot cell. When present, chlamydospores are intercalary, single, in chains or clumps and are globose, thick-walled, hyaline to pale-brown with a smooth or slightly roughened outer wall; they are 10 to 12 µm in diameter. Many strains fail to develop chlamydospores on standard media (Booth, 1973).

Teleomorph: Ascomata are perithecioid and are superficial on a thin stroma forming in clusters around the lower nodes or base of infected stems. They are ovoid, 140 to 250 µm diameter, with a rough tuberculate outer wall; they show varying degrees of lateral collapse when dry. The ascomatal wall consists of two layers, an outer stromatic layer, 17 to 31 µm wide, comprising globose cells, which are 5 to 12 x 1.5 to 3.5 µm, and a thin inner layer of compressed thin-walled cells. Asci are 60 to 85 x 8 to 11 µm, clavate, with a short stipe and eight or occasionally 4 to 6 distichous or obliquely monostichous ascospores. Ascospores are hyaline to light-brown, curved fusoid with rounded ends, initially 0 to 1-septate and finally 3-septate, 19 to 24 x 3 to 4 µm.

Biology and Ecology

The primary inoculum source of *G. zeae* is infested crop residues, from which ascospores and conidia are released (Windels and Kommedahl, 1984). However, comparison of conidial- and ascospore-derived disease gradients in wheat by Fernando et al. (1997) suggested a lack of secondary infection, confirming that *G. zeae* head blight is primarily a monocyclic disease in this crop. In field observations in China, mature perithecia were found throughout the year. Perithecia could germinate on a wide range of crop residues, including rice, wheat and maize.

During 1973 to 1983, 14.6% of nearly 7500 green maize stalks, collected from mid-September to early November in Minnesota were infected by *G. zeae*. Of rotten (standing or lodged) stalks collected at the same time as green stalks in 1981 and 1982, 84.4% yielded *G. zeae.* Recovery from overwintered, standing maize fields the following spring was 61%, and recovery from 1-year-old basal stalk pieces the following autumn was 31%. Mature perithecia developed in 99% of the isolates tested. The pathogen colonizes pith tissue late in the growing season as stalks ripen, and predominates as tissues senesce, thereby increasing its inoculum potential and survival niches for the following season (Windels and Kommedahl, 1984). *G. zeae* was commonly isolated from soybean stems and pods during a two year study of fungal survival in a no tillage system (Baird et al., 1997).

F. graminearum also survives as hyphae in large pieces of maize stubble derived from stalks colonized parasitically or saprophytically during senescence, and as hyphae in small pieces of organic debris (Wearing and Burgess, 1978). Another potential source of inoculum is infected seeds (Duthie and Hall, 1987). The pathogen also may be transmitted by birds and insects (Sutton, 1982). Adult female wheat midges (*Sitodiplosis mosellana*) have been shown to transfer spores of *G. zeae* from infected to non-infected wheat spikes (Mongrain et al., 2000).

Formation of *G. zeae* ascomata occurs at 5 to 35°C and ascospore production at 13 to 33°C (optimum 25 to 28°C). The former is favored by diffuse light, especially in the early stages, but asci and ascospores may be produced in darkness. More ascospores are released at night than by day and on rainy days. Spore discharge peaks between 2200 and 0800 hours. Ascospores germinate at 4 to 35°C (optimum 25 to 28°C). Over 90% germinate within 4 to 8 hours when incubated at 25 to 30°C. Germination is inhibited by <81% relative humidity (RH) but does not require light or exogenous nutrients (Ye, 1980). Daily average densities of macroconidia of the anamorph were an order of magnitude less than ascospores.

Infection of wheat, barley and maize by *G. zeae* is influenced by temperature and moisture. In Croatia, scab of wheat is favored by high temperatures (25 to 30°C) and high relative humidity (above 85% RH) (Tomasovic et al., 1993). The occurrence of scab on wheat and barley in Japan was related to the amount of precipitation and temperatures in April and May, and the time of ploughing of the paddy fields where inoculum survived on rice stubble, according to the analyses of information obtained from 1955 to 1980. Weather records from 1889 to 1980 indicated that scab incidence increased with higher spring temperatures and a greater number of days with more than 5 mm rain in May. The most severe epidemics were reported in 1890, 1923 and 1963, and these were correlated with favorable weather for infection during April (CABI, 2004). Physical injury to maize ears by insects, and infection through the silk on the ear tip, were found to be the major routes of fungal entry by *G. zeae* in the development of ear rot in Ontario (Vigier et al., 1997).

Pest Importance

Many plant species are affected by *G. zeae*, but those of major economic importance include maize, and small grains, particularly wheat, barley, rye and triticale. The fungus attacks the cob or cereal spikes resulting in pink ear rot of maize, lightweight, chalky-white Fusarium damaged kernels (FDK) of wheat, and shrunken discolored kernels of other grains. Losses are not restricted to yield and quality, however, but are incurred throughout the grain industry by millers, bakers, pasta makers, maltsters, brewers and feedlot operators (Gilbert and Tekauz, 2000; Tekauz et al., 2000). In addition to yield loss, Gibberella ear rot causes the production of mycotoxins in diseased ears and kernels that contaminate animal and human food. Of particular concern are vomitoxin that

causes a vomiting syndome, zearalenone that causes hyperoestrogenism and the tricothene toxin, T-2-toxin. Surveys have shown that these mycotoxins cause extensive contamination of maize and wheat grain throughout the world (Tuite et al., 1990). A survey of commercial small cereal grains in Minnesota, North Dakota and South Dakota in 1993 to 1994 by Jones and Mirocha (1999) detected deoxynivalenol (DON) [vomitoxin] in 493 of 500 samples of wheat, 100 out of 100 six-row barley samples and in 28 samples of oats. In an evaluation of semolina milling and pasta-making quality in ten durum wheat cultivars harvested in Manitoba, Dexter et al. (1997) found that the retention of vomitoxin in the semolina was approximately 50% and the semolina yield was lower than in unaffected grain.

Fusarium head blight (FHB) epidemics are sporadic in nature requiring rain or high humidity at flowering, in addition to the presence of susceptible hosts and inoculum. In recent decades, years with severe losses due to FHB have been numerous, and regions recording severe losses appear to have increased. Fusarium head blight, or scab, is not a new disease in the U.S. In 1917, 31 of 40 states that were surveyed reported damage from FHB with losses estimated at 288,000 metric tons, primarily from the winter wheat areas of Ohio, Indiana and Illinois. In 1919, losses caused by the disease were estimated at 2.18 million metric tons throughout the U.S. (McMullen et al., 1997). A major epidemic affected 4 million hectares of the spring wheat and barley growing area of the northern Great Plains of North and South Dakota and Minnesota. Yield losses exceeded 6.5 million tons worth $826 million dollars, although total losses associated with the epidemic approached one billion dollars. In subsequent years, losses in these states have been estimated at $200 to 400 million annually (McMullen et al., 1997). Losses in barley due to FHB are largely due to the presence of DON. In 1996, barley prices in Minnesota dropped to $2.75 from $3.00 per bushel if the mycotoxin was detected and an additional $0.05 for each additional ppm DON detected. Malting barley was sold at feed prices, as low as $2.25 per bushel (CABI, 2004).

In Argentina, soybean crop residues in fields under conservation tillage have been found to be heavily infested with *F. graminearum (*Pioli et al., 2004). Although the fungus also reportedly grows on living soybean stems and seeds, many consider the fungus non-pathogenic to soybean. *F. graminearum* was isolated from stems and seeds of symptomatic soybean plants and found to be pathogenic on soybean after completion of Koch's postulates in Argentina (Pioli et al., 2004). Recent surveys of soybean seed grown in Brazil also revealed infection by *F. graminearum*. *F. graminearum* strains from

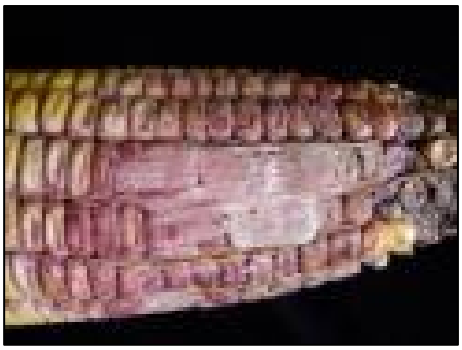


Figure 2. Symptoms of Gibberella ear rot. Photo courtesy of 1710Hwww.saspp.org

Brazilian soybean seed consistently caused pod rot and root rot disease on all soybean varieties, under all conditions tested. These same strains also caused FHB in wheat (Martinelli et al., 2004). Two strains from *F. graminearum* lineage 7 from the U.S. also caused symptoms on soybean (Martinelli et al., 2004). Farmers who use a soybean/wheat crop rotation should be aware of a potential build-up of strains that infect both wheat and soybean, reducing the effectiveness of the rotation. Brazilian strains, which also produce a novel mycotoxin known as 3-acetylnivalenol, have not yet been found in the U.S.

Symptoms/Signs

Soybean: Stems often have a brown discoloration that may extend progressively along the stem. Interveinal chlorosis or loss of turgence of unifoliate leaves and interveinal chlorosis of trifoliate leaves followed by plant wilting and death are also common. Roots with light brown or necrotic areas (Fig. 3). Pods developed large (>1 cm) dark brown, necrotic lesions. Younger pods blighted and dropped from the plant (Piolo et al., 2004; Martinelli et al., 2004).

Maize: Leaves on early-infected plants suddenly turn a dull greyish-green; while the lower internodes soften and turn tan to dark-brown is a characteristic symptom of Gibberella stalk rot. Diseased tissue within the stalks often shows a pink to reddish discoloration. The fungus causes shredding of the pith and may produce small, round, black perithecia superficially on the stalks. Lesions may develop concentric rings. A reddish mold, often at the

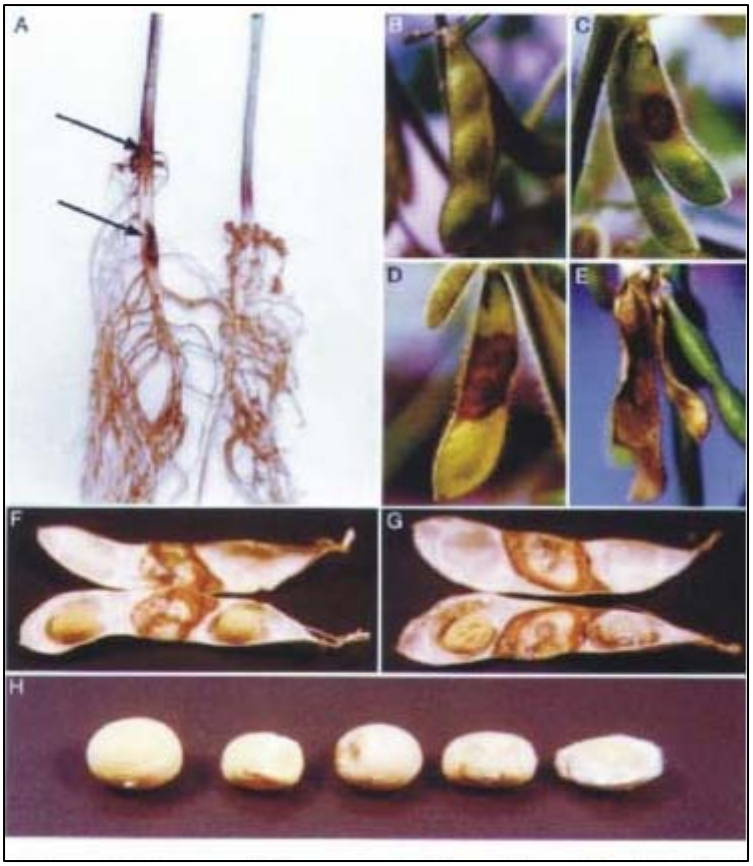


Figure 3. Symptoms of disease caused by *F. graminearum* on soybean. A) (left) lesions on crown and hypocotyl of soybean (arrows) grown in soil infested with *F. graminearum*, (right) non-symptomatic soybean grown in non-infested soil, B) control pod, C) inoculated pod showing discrete, chocolate-brown lesions, D) a dry, tan to brown, expanding pod lesion, E) blighting of young inoculated pods, F) internal symptoms of pod showing necrosis limited to the dentral inoculated carpel, G) internal symptoms of pod showing spread of infection to adjacent seed, H) seed from inoculated pods ranging from non-symptomatic (left), to a brown necrotic spot (center), to largely colonized by fungal hyphae (right). Photos courtesy of Martinelli et al., 2004 Fitopathologia Brasileira 29: 492-498.

ear tip, is the characteristic sign of Gibberella ear rot (Fig. 2). Early infected ears may rot completely, with the husks adhering tightly to the ear and a pinkish to reddish mold growing between the husks and ear.

Sorghum: *G. zeae* can affect sorghum at all growth stages. Lesions vary in size from small, circular spots to elongated streaks. They may be light-red to dark-purple. Lesions may be found in the interior and on exterior tissues of roots, stalks, seeds and peduncles. Dark red discoloration of the cortex of seedling roots is often observed, and the fungus may spread to other root and stalk tissue during the growing season. In seedlings and young plants, leaves turn brown and the plants wither and die; under very humid conditions whitish-yellow mycelium develops, which later becomes salmon-pink. In older plants, the pathogen invades the vascular bundles and inner tissues of the stalk which then become reddish. Early-infected flowers or young grain may be destroyed; mature grains may become covered with mycelium, but are not destroyed (Tarr, 1962).

Figure 4. A healthy wheat head (left) stands in contrast to one inoculated with *F. graminearum* showing severe symptoms of Fusarium head blight disease (right). Photo courtesy of USDA-ARS Cereal Disease Laboratory.

Rice: *G. zeae* may produce a reddish appearance on affected seeds. Discoloration of definite areas of the seed, appearing as brown spots, or covering the entire surface of the seed, may also occur. The fungus causes the formation of spots on the surface of the husks which are at first white, but later become yellow and salmon or carmine. Infected grains are light, shrunken and brittle. Nodes of stems are attacked, causing them to rot, turn black and disintegrate. Stems wilt, break and lodge (Padwick, 1950).

Figure 5. Pink color on wheat spike due to spore masses of *F. graminearum*. Photo courtesy of G. Bergstrom, Cornell University.

Wheat: Blighted seedlings are characterized by a light-brown to reddish-brown water-soaked cortical rot and blight before or after emergence. Head blight is conspicuous before the spikes mature. Infected spikelets first appear water-soaked; this is followed by the loss of chlorophyll, giving a final bleached straw color (Fig. 4).

During warm, humid weather, conidial development is abundant and the infected spikelets show a pink or

salmon-pink cast, especially at the base and in the crease of the kernel (Fig. 5). For wheat, brown, dark purple to black necrotic lesions form on the exterior surface of the florets and glume. Infection may spread to adjacent spikelets or through the entire spike. The infected kernels become shriveled, with a scabby appearance due to the tufty mycelial outgrowths from the pericarp. Although these lesion symptoms sometimes are referred to as scab, they are not formally related to the hyperplasia and hypertrophic epidermal growth associated with other scab diseases such as apple scab. Infected kernels range in color from white to pink to light-brown, depending upon the time of infection and environmental conditions during disease development (Dickson, 1947). Peduncles immediately below the inflorescence may become discolored brown/purple. With time, tissue of the inflorescence often becomes blighted, appearing bleached and tan, while the grain within atrophies. Awns often become deformed, twisted and curved downward.

Barley: Infections are not always readily apparent in the field. Restricted, reddish-brown cortical lesions occur when infected seed is sown in cool, moist soil. In warm soil, seedling blight may occur before or after emergence. During later stages of plant development, crown and basal culm rot are commonly observed. Spikes are dwarfed and compressed with infected spikes closed rather than spread. All or part of the spike is infected (Fig. 6). Hulls (lemma and palea) are light to dark-brown with a dead, lustreless surface. Conidial or perithecial masses commonly develop on the surface, especially during moist weather. Kernels are shrunken and light brown in color. The pericarp surface is rough or scabby in appearance.

Figure 6. Barley with symptoms of Fusarium head blight. Photo courtesy of B. Steffenson, J. Pederson, and V. Pederson, North Dakota State Unversity.

Known Hosts

Major hosts

Alopecurus pratensis (meadow foxtail), *Avena sativa* (oats), *Glycine max* (soybean), *Hordeum vulgare* (barley), *Linum usitatissimum* (flax), *Lupinus* (lupine), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Pennisetum glaucum* (pearl millet), *Secale cereale* (rye), *Sorghum bicolor* (sorghum), *Triticum aestivum* (wheat), and *Zea mays* (maize).

Minor hosts

Acacia mearnsii (black wattle), *Azadirachta indica* (neem tree), *Brassica* spp., *Dianthus* spp. (carnation), *Gardenia jasminoides* (cape jasmine), *Gossypium* spp. (cotton), *Lycopersicon* spp., *Mangifera indica* (mango), *Medicago* spp. (medic), *Musa* x *paradisiaca* (plantain), *Panicum miliaceaum* (millet), *Phaseolus vulgaris* (common bean), *Pinus sylvestris* (Scots pine), *Pisum* spp.(pea), *Rubus ideaeus* (raspberry), *Solanum* spp. (nightshade), *Trifolium* spp.(clovers), *Triticale* spp., *Vicia faba* (broad bean), and *Zingiber officinale* (ginger).

Known Distribution

The disease, caused by *G. zeae*, is prevalent in areas with continental climates such as parts of Asia (China, Japan), North and South America (Canada, U.S., Mexico, Uruguay, Argentina) and Europe (Bulgaria, Germany, Hungary, Italy, Poland, Romania, Russia, Yugoslavia). In temperate or maritime regions the disease is usually caused by *Fusarium culmorum*. Other countries that have reported either *F. graminearum* or *G. zea* include: **Asia:** India, Iran, Kazakhstan, Korea, Lebanon, Pakistan, Saudi Arabia, Sri Lanka, Turkey. **Europe:** Austria, Croatia, Finland, former Czechoslovakia, France, Greece, Iceland, Ireland, Lithuania, Moldova, Netherlands, Norway, Portugal, Slovakia, Slovenia, Spain, Sweden, Switzerland, Ukraine, United Kingdom. **Africa:** Egypt, Gambia, Kenya, Malawi, Nigeria, South Africa, Tunisia, Zimbabwe. **Central America:** Costa Rica, Dominica, Grenada, Honduras, Saint Lucia, Saint Vincent and the Grenadines, Windward Islands. **South America:** Bolivia, Brazil, Colombia, Paraguay, Peru. **Oceania:** Australia, Fiji, New Zealand, Papua New Guinea, and Solomon Islands.

Potential Distribution Within the US

F. graminearum is currently present in California, Colorado, Connecticut, Florida, Georgia, Hawaii, Idaho, Illinois, Indiana, Iowa, Kansas, Louisiana, Michigan, Minnesota, Mississippi, Missouri, Nebraska, New York, North Carolina, North Dakota, Ohio, Oklahoma, Oregon, South Dakota, Tennessee, Texas, Washington, and Wisconsin on cereal crops and grain. However, the potential distribution of strains pathogenic on soybean within the U.S. is currently unknown.

Survey

Survey for *F. graminearum* and *G. zea* is primarily based on symptom observation during a visual survey.

In maize, root rots and seedling blight occur. Look for dull, greyish-green color on early leaves of infected plants, and brown to black lesions, in which black ascomata may occur, near lower nodes. The pith is shredded and pink-to-red. A red mold begins at the tip, and may spread to the whole ear. Black ascomata can form on the seeds and husk. Look for small, round, black ascomata on the stalk. This disease can be differentiated from Diplodia stalk rot by pith discoloration and the superficial ascomata. Superficial blue-black ascomata are sometimes found on husks and ear shanks.

In sorghum, a dark red discoloration of the cortex of seedling roots is often observed as an early season symptom. Premature plant death, usually during grain development, is often a diagnostic characteristic for Fusarium stalk rot. Look for plants whose leaves suddenly turn bluish-grey, closely resembling frost damage or sun scorch; the exterior stalk tissue usually remains green. Look for ascomata which appear on the dead stalks of older plants. Mature grains

become covered with mycelium, but are not destroyed. Under very wet conditions, look for dense, whitish-pink mycelium covering an affected inflorescence. Later, small, blue-black ascomata are visible to the naked eye (Tarr, 1962).

In rice, look for bleached lesions or a bleached discoloration on the glumes; affected areas become yellow to salmon or carmine. Infected grains are light, shrunken and brittle, and may be sterile. *G. zeae* may infect the nodes of tillers, causing a black rot and resulting in the breakage of stems. Sporodochial conidiomata, conidial masses and blue-black ascomata may be observed on infected glumes. Ascomata are formed at the nodes of infected stems.

In wheat, scab is best recognized on emerged immature heads where one or more spikelets or the entire head appears prematurely bleached. If the rachis is infected, all tissues above that point will be faded. Look for tufty mycelial outgrowths from the pericarp of infected kernels. Small, dark ascomata and superficial (frequently pink or orange) mycelium and spore masses may be seen on, and especially at the base of, diseased spikelets.

In barley, look for infected developing kernels which are usually shrunken, and greyish-brown, especially towards the base of the spikelet. The interior of infected kernels becomes floury with a greyish discoloration. A salmon-pink to reddish, fluffy, dust-like mycelial growth is frequently evident along the edge of the glumes or at the base of the spikelet. If warm, moist weather persists, blue-black ascomata may be observed on early-infected heads by harvest time.
In soybean, look for a brown discoloration on the stem, interveinal chlorosis on leaves, and root rot.

Key Diagnostics

F. graminearum may be cultured on selective media (Kanatani and Takeda, 1991; Szecsi and Mesterhazy, 1998). Conventional and competitive polymerase chain reaction (PCR) assays have been developed to detect and quantify *G. zeae* in wheat heads (Nicholson et al., 1998). PCR analysis has been compared with visual assessment of FHB of wheat (Doohan et al. 1998). Near-infrared spectroscopy (NIRS) has been used to detect FHB damage caused by *G. zeae* and estimate deoxynivalenol (DON) and ergosterol levels in single wheat kernels (Dowell et al., 1998). An excellent new reference for Fusarium is: Fusarium Laboratory Manual. Leslie, J.F. and Summerell, B.A., 1st Ed., 2006, Blackwell Publishing, pp. 388.

Passalora sojina

Scientific Name
Passalora sojina (Hara), H.D. Shin & U. Braun

Synonyms:
Cercospora sojina, *Cercospora daizu* miuri, *Cercosporidium sojinum*

Common Name(s)
Frogeye leaf spot, frogeye disease, grey speck, leaf spot of soybean.

Figure 1. Fascicles of *P. sojina* on seed. Photo courtesy of J. T. Yorinori.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota **Class:** Ascomycetes **Order:** Mycosphaerellales, **Family:** Mycosphaerellaceae

Reason for Inclusion in Manual

Notes on Nomenclature
Passalora sojina is the new proposed name for *Cercospora sojina*. The genus *Passalora* includes *Cercospora*-like species with slightly thickened conidial scars and subglose, ellipsoid-ovoid or broadly obclavate-fusiform, sparsely septate conidia, as well as species with obclavate-fusiform, pigmented, multiseptate conidia.

Pest Description
A detailed description of *P. sojina* was given by Chupp (1953). Stromata are lacking or are small and brown. Conidiophores are borne singly or in dense fascicles (Fig. 1), sparingly septate, pale brown, uniform in color, slightly attenuated, straight to sinuous, 1 to 12 geniculations (average 1 to 3), not branched, medium-sized spore scar at subtruncate to rounded tip, and 4 to 6.5 x

40 to 200 µm in size. Conidia (Fig. 2) are hyaline, cylindric to cylindric-obclavate, rarely acicular, straight to mildly curved, septa from 3 to 13, base subtruncate to obconically truncate, tip obtuse, 4 to 8 x 20 to 80 µm, rarely 120 µm. Size of conidia depend on the isolate, race or the substrate on which it is produced. *P. sojina* develops a thin, dark-grey to black mycelium on potato-dextrose agar. Some isolates show white mycelium on a dark background or develop tufts of white mycelium on dark colonies.

Biology and Ecology

P. sojina survives on infected plant residues and seeds. These are the primary sources of inoculum for the following season and for the establishment of the disease in new fields (Sherwin and Kreitlow, 1952). Soybean seedlings from infected seeds develop lesions on the cotyledons and generate the early infections on leaves of adjacent seedlings grown from healthy seeds. From a source of inoculum, conidia are easily windborne and wind dispersed. The temperature range for infection is 15 to 32°C, with the optimum of 25 to 28°C. Conidia of *P. sojina* germinate within 1 hour in water but require a film of water on the leaf surface for at least 6 hours and 48 hours of high moisture for an adequate disease development. Frequent rains following the onset of disease are required for epidemics to occur. Disease development in the field seems to be hindered when night temperatures fall below 20°C. Prolonged periods of leaf wetness and cloudy days reduce epidemics, because the conidia germinate before they are windborne.

In most susceptible cultivars, the first pod symptoms appear when the seeds are about half of their full size. An early pod infection leads to a greater damage and rate of seed transmission. Pod infection by *P. sojina* may increase, pod and seed infection by *Cercospora kikuchii*, *Colletotrichum truncatum*, *Diaporthe phaseolorum* var. *sojae,* and *Fusarium semitectum*; however, recovery of *P. sojina* from stems, pods, and seeds is not affected by the presence of other fungi (Bisht and Sinclair, 1985).

P. sojina develops through the pod wall to reach the developing seed, and penetrates the seed coat through pores, cracks, or the hilar region. It forms a thick brown hyphal mat in the parenchymatous region of the seed coat, and lysis the nearby cells. The pathogen is rarely found in other tissues of the seed coat (Kunwar et al., 1985). The colonized seed coat remains attached to the

Figure 2. Conidiophores and conidia of *P. sojina*. Photo Courtesy of J. T. Yorinori.

cotyledon until the seedling has fully emerged and the conidia in the seed coat is readily available for wind dispersal.

Best control of frogeye leaf spot results by growing resistant cultivars. Most races of the pathogen have a wide geographical distribution and the same cultivar may be the host for several races. Resistance to race 1, in cultivars Lincoln and Wabash, and resistance to race 2, in cultivar Kent, are controlled by major dominant genes, designated Rcs1 and Rcs2, respectively (Probst and Athow, 1958; Probst et al., 1965). Resistance to race 5, in cultivars Lincoln and Davis, is conditioned by two independent dominant genes at different loci (Phillips and Boerma, 1982). The gene in cultivar Davis was designated Rcs3 (Boerma and Phillips, 1983). The Rcs3 gene is believed to condition resistance to all known races of *P. sojina* (Phillips and Boerma, 1982).

Because *P. sojina* does not significantly affect seed germination, it is not regarded as an important seed pathogen, but the primary objective of seed treatment with fungicides is to prevent the introduction of the fungus, or a new race, into a new field. Another benefit of seed treatment in tropical and sub-tropical regions is to ensure the germination of less vigorous seed in soils with low moisture. Since the first symptoms of the disease normally appear at early bloom, if the weather is favorable for *P. sojina*, the first application of fungicides should start when the most infected leaflets, on randomly sampled plants, have an average of 5 to 10 spots. Control measures such as plowing under crop residues, 2 to 3-year rotation, and use of early-maturing cultivars have also been recommended.

Figure 3. Symptoms of frogeye leaf spot. Photo Courtesy of X. B. Yang.

Pest Importance

P. sojina is distributed worldwide. Thus, quarantine regulations to restrict its movement are not justified. Nevertheless, it is important to take precautionary measures to prevent the introduction of new races from other regions or countries.

P. sojina is capable of causing significant yield reductions in warm and humid conditions (Phillips, 1999), especially in late-maturing cultivars. Yield losses in the U.S. varied from 17 to 21% during 1966 to 1968 (Laviolette et al., 1970). Since 1952, frogeye leaf spot has decreased in importance and almost disappeared in the Midwestern states of the U.S. However, an outbreak of the

disease occurred in Iowa in 2001 (Yang, et al., 2001). The decrease in the incidence of the disease was attributed to the use of resistant cultivars and unfavorable weather conditions. It is still common in the southern states, causing occasional damage on susceptible cultivars. Frogeye leaf spot causes severe damage in Brazil (crop losses of 100%), Nigeria, Zimbabwe, and China.

Symptoms/Signs

First visible symptoms appear as white-green, water-soaking or wilting spots, evolving to grayish-green, circular to subcircular, varying in size from minute spots to 5 mm in diameter. Individual lesions may coalesce to form large, irregular lesions. The difference in lesion color between the lower and upper leaf surfaces is the key feature for diagnosing frogeye leaf spot in the field. On the upper leaf surface, as the lesions become older, necrotic tissue progressively turns reddish-brown, light-brown and finally paper-white (Fig 3). Fully developed lesions may present an outer thin yellow halo surrounding a reddish-brown margin and a reddish-brown to light-brown center. On the lower leaf surface, the lesion color varies from light-green to grayish-green and finally to light-brown, usually with black tufts due to the formation of conidiophores and conidia, which may occur 24 hours after the lesions are first visible. Lesions formed on leaves in the shaded portion of the lower canopy may have sporulation on both sides.

The youngest fully developed leaves at the time of infection with *P. sojina* will show the greatest number of lesions. Leaf spots are larger on younger leaves and are smaller in size as the plant grows towards maturity. There is no progressive increase in lesion size once the lesions become visible, but the color of the lesions changes. Severe infections by *P. sojina* result in early defoliation, poor grain formation, and uneven maturation.

On the stem, lesions are mainly confined in the cortex, and start as small (<1 mm diameter) reddish-brown spots and enlarge to an elliptical shape more than 1 cm long. Older lesions have a thin dark reddish-brown margin and progressively lighter color in the center. Under favorable environmental conditions for sporulation, a dark-grey layer of conidiophores forms in the center of the lesion.

The development of lesions on the pods is similar to that on the leaves. Symptoms are first visible as water-soaked, circular or slightly circular spots of various sizes (1 to 5 mm in diameter), which later become light-brown in the center. Under heavy infections, lesions may coalesce and turn dark-grey, resulting in rotting of the pod and seeds.

Seeds infected by *P. sojina* have conspicuous light to dark-grey or brown areas ranging from minute specks to large blotches covering the entire seed coat. Some lesions show light and dark brown alternating bands (Sherwin and Kreitlow, 1952). Infected seeds usually show cracking of the seed coat (Phillips, 1999). Seed damage depends on cultivar susceptibility and the growth stage

when pod infections occur. Early defoliation causes smaller seeds with green seed coats. Early infected pods may have completely rotten seeds.

Known Hosts

Major host
Glycine max (soybean)

Minor host
Mucuna (velvet beans)

Known Distribution

P. sojina is distributed worldwide. It is more prevalent and destructive in warm and humid tropical and sub-tropical regions. CABI (2004) lists the countries where frogeye leaf spot has been found: **Asia:** China, India, Indonesia, Japan, Korea, Nepal, Timor, Vietnam, Latvia, and the USSR. **Africa:** Cameroon, Ivory Coast, Egypt, Gabon, Kenya, Malawi, Nigeria, Zambia, and Zimbabwe. **North America:** Canada, Mexico, and the U.S. **Central America:** Cuba and Guatemala. **South America:** Argentina, Bolivia, Brazil, and Venezuela. **Oceania:** Tonga.

Potential Distribution Within the US

The disease is present in: Alabama, Arkansas, Delaware, Florida, Georgia, Hawaii, Illinois, Indiana, Iowa, Kansas, Louisiana, Maryland, Michigan, Mississippi, Missouri, New Jersey, New York, North Carolina, Oklahoma, South Carolina, Texas, Virginia, West Virginia, and Wisconsin.

Survey

Survey for frogeye leaf spot is based on visual survey for symptoms. The first leaf symptoms are generally observed from the early flowering stage. Stem and pod symptoms usually appear and progress from the later stage of pod filling (growth stage R5). The more susceptible the cultivar, the earlier the pod and stem symptoms will be expressed, but none are earlier than the half-pod-filling stage (R5).

Leaves: Circular to sub-circular leaf spots, varying in size from minute reddish-brown dots to 5 mm diameter lesions. A diagnostic trait for frogeye leaf spot is the difference of color of lesions on the upper and lower leaf surfaces. On the upper leaf surface, fully developed lesions have a reddish-brown margin and a lighter brown centre (Fig. 3). On the lower leaf surface, the lesions vary from grey to tan, depending on the intensity of sporulation.

Pods: Water-soaked, circular or slightly circular spots (1 to 5 mm diam.), later, light-brown in the center.

Seeds: Grey to brownish areas, ranging from minute specks to large blotches covering the entire seed coat.

Key Diagnostics

P. sojina is easily diagnosed if conidia are present on fresh infected leaves or after 24-hour incubation in a moist chamber. It produces abundant conidia on all infected plant parts and on crop residues. Conidiophores and conidia are scraped from the lower leaf surface onto a drop of water on a glass slide and examined under a microscope under 100 to 500x magnification. An alternative procedure for spore collection consists of using transparent sticky tape as a substitute for the cover glass. A loop made up of a 3 to 4-cm long piece of tape is lightly touched (sticky side) on the sporulating lesion. The tape is then mounted on a glass slide by stretching the tape with the surface having the spores facing a drop of water. The slide is examined for the presence of conidia under a microscope.

Diagnosis of frogeye leaf spot if the leaves are no longer available can be accomplished by collecting stem pieces and pods with lesions (Yorinori, 1980). The stem pieces are incubated in a moist chamber for 24 to 48 hours to allow sporulation. Infected pods should be carefully collected to prevent shattering. The pods are surface disinfested for 3 to 4 seconds in 95% ethanol, followed by 3 to 4 min. in 1% sodium hypochlorite, and then washed under running tap water. Seeds are aseptically removed from pods, plated (10 seeds/9 cm plate) on filter paper, and incubated at 25 ± 2°C. Seeds from the pods can also be plated (5 seeds/plate) on potato dextrose agar (PDA) or other suitable media for isolation.

The blotter method suggested by the International Seed Testing Association may also be used to detect *P. sojina* in seeds. An incubation chamber (9 cm diameter Petri dish) with four layers of moist filter paper is used. Ten to fifteen seeds are placed in the dish and incubated for 4 to 5 days at room temperature (25 ± 1°C). A 12-hour light/dark cycle may enhance sporulation. The incubation period should not exceed 5 days; conidia tend to germinate readily once they are mature and make identification difficult. Disinfestation of seeds with sodium hypochlorite before plating may be helpful to reduce the surface fungal contaminants without affecting the recovery of *P. sojina* (Grybauskas et al., 1979).

After the incubation period, each stem piece or seed should be examined for the presence of *P. sojina* using a stereomicroscope at 60 to 100x magnification. Typically *P. sojina* has straight to mildly curved hyaline cylindric to cylindric-obclavate conidia (Fig. 2).

Frogeye leaf spot may be confused with the early stages and old lesions of leaf spot caused by the fungus *Myrothecium roridum*, a common soybean pathogen in the tropics. Necrotic lesions caused by spray drift of some herbicides and

downy mildew (*Peronospora manshurica*) may be also confused with frogeye leaf spot.

Differentiation from Asian soybean rust

Lesions of Asian soybean rust are clustered alongside the veins and have pustules with pores at the top of the cone and masses of uredionospores, whereas, lesions of frogeye leaf spot do not have pustules. Lesions of frogeye leaf spot are larger and have distinct purple to reddish-brown margins (Fig. 3).

Peronospora manshurica

Scientific Name
Peronospora manshurica (Naumov) Syd. ex Gäum.

Synonyms:
Peronospora sojae, *Peronospora trifoliorum* var. *manshurica*

Common Name(s)
Downy mildew, soybean downy mildew

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Oomycota, **Class:** Oomycetes, **Class:** Peronosporales, **Family:** Peronosporaceae

Reason for Inclusion in Manual

Pest Description
According to Francis (1981), *P. manshurica* develops an intercellular mycelium and filiform twisted haustoria in the host cells. Sporangiophores (Fig. 1) are 350 to 800 x 6 to 8 µm in size; the length of the trunk section is between 200 and 600

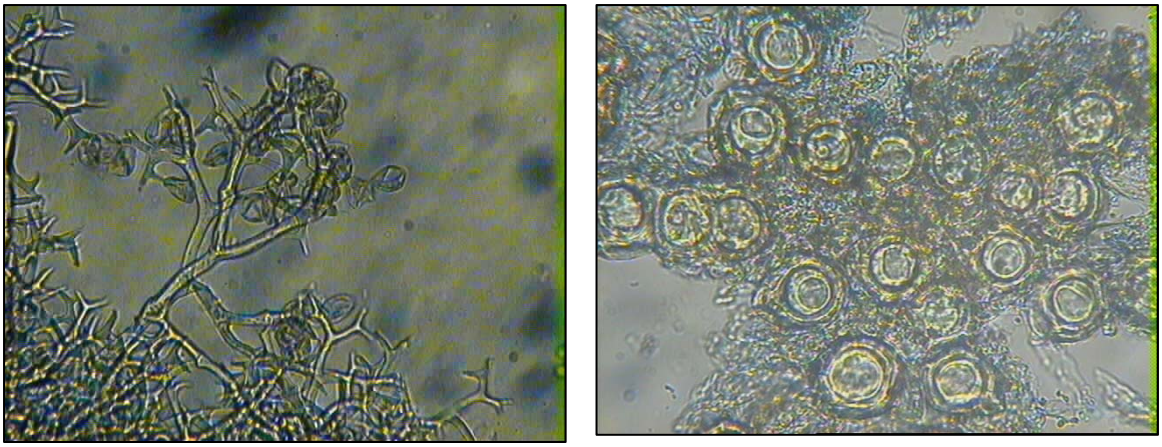


Figure 1. Sporangiophores and sporangia (left) and oospores of *P. manshurica* (right). Photos courtesy of Azucena Ridao

µm; branching is indistinctly dichotomous, 4 to 5 times; branch ends widely reflexed, often forming a right angle, short (6 to 8 µm) and abruptly tapered giving a rather stiff appearance. Sporangiospores (conidia) are 20 to 26 x 18 to 21 µm in size, ellipsoid to near globose, and do not produce zoospores. Oospores (Fig. 1) are 30 to 50 µm in size, its outer wall is pale yellow and will eventually become reticulated (Phillips, 1999); they occur in large numbers in seeds and leaves of systemically infected plants (Francis, 1981). They are also found in the pith tissue of petioles, stems, and roots.

Biology and Ecology

P. manshurica is a seed transmitted pathogen. Seedlings are infected while developing from oospore-encrusted seeds (Hildebrand and Koch, 1951). Infected seedlings also develop if healthy seeds had been mixed with infected pod tissues and stored over the winter in the warehouse (Hildebrand and Koch, 1951). Early infected plants on which sporulation occurs are the inoculum source for other plants in the field (McKenzie and Wyllie, 1971). Free water on leaf surface favors conidial germination and infection. The disease is favored by high humidity and temperatures of 20 to 22ºC. Under these conditions, the pathogen can spread rapidly (wind-borne) from the primary center of inoculum and infect 100% of the plants by the end of the growing season. No sporulation occurs above 30ºC or below 10ºC. When *P. manshurica* invades the pod, the hyphae extend through the parenchyma and the endocarp, and form oospores. Pods borne in branches from the 3rd and 4th node are often the most severely infected. In systemically infected plants, mycelium, antheridia, oogonia and oospores of the pathogen line the inner pod wall, and cover the seed coats. In addition to oospores on the seed coat, considerable mycelium is present within the palisade and hour-glass cells. The fungus is much less apparent in the embryo. It is absent in the hypocotyl or plumule, and only rarely present in the cotyledonary tissue (Hildebrand and Koch, 1951). The encrustation on the seed coat includes oospores, oogonia, gall like thick-walled hyphae, and sclerotia-like structures. A lesser amount of thin-walled hyphae are also present. The oospores are attached to the seed coat by hyphal connections. (Roongruangsree et al., 1988a).

Figure 2. Symptoms of downy mildew on the upper leaf surface. Photo Courtesy of Clemson University-USDA Cooperative Extension Slide Series. 1711Hwww.invasive.org

P. manshurica has several races. Three were identified in 1950 (Geesman,

1950). Surveys in the U.S., indicated that the prevalence of various races between 1971 and 1976 changed continuously (Dunleavy 1977). To date, at least 35 races have been described, and more than one race may be present in one plant.

A survey of seed samples in the U.S., showed that 73% of seed lots contained oospore-encrusted seeds with an incidence in seeds of 25%. Infected seeds were lighter and smaller (McKenzie and Wyllie, 1971), and reduced or delayed the emergence in the field (Hildebrand and Koch, 1951). Plants grown from infected seeds had reduced height, less seed set, less 1000 seed weight, and less seed protein (Koretsky and Koretsky, 1998).

To control the disease, cultural practices such as planting pathogen-free seed, crop rotation, and avoiding cultivation when foliage is wet are recommended. Treatment of plants with fungicides may decrease the numbers of oospore-encrusted seeds in susceptible cultivars (Dunleavy, 1987). Numerous resistant cultivars have been released throughout the world. Cultivars with the *Rpm* gene are resistant to races 1 to 32 but susceptible to race 33 (Phillips, 1999). The resistance conferred by gene *Rpm* has been associated with a hypersensitive response (Ersek et al., 1982).

Pest Importance

Downy mildew rarely causes significant economic losses, but the disease is widely distributed. For example, in 1984, it was found in 50% of 825 fields surveyed in Iowa (Dunleavy et al., 1984). Jones and Torrie (1946) reported that the productivity of plants growing from infected seeds was decreased by 6%. In contrast, Hildebrand and Koch (1951) did not find significant differences in yield between plants grown from encrusted or healthy seeds. In another study, a yield reduction of 11.8% was noted in susceptible cultivars, but this yield reduction was absent in resistant cultivars (Dunleavy, 1987).

Symptoms/Signs

Downy mildew is primarily a foliar disease, but seeds, pods, and stems can also be infected. Younger leaves are more susceptible to downy mildew than older leaves. Small, discrete, pale to bright yellow spots (2 to 8 mm diameter) are formed on the upper leaf surface (Fig. 2). Under severe infections, the entire leaf area may be infected, shrivel and die early (Francis, 1981). Prolific production of sporangiophore can occur on the undersurface of leaves. These sporangiophores form a

Figure 3. Symptoms of downy mildew on the lower leaf surface. Photo courtesy of David Faulkner.

grayish-purple down beneath the spots (Fig. 3). Pods can become infected without showing external symptoms, and the seeds invaded. Oospores develop on the seed surface and appear as a milky-white crust made up of a mass of the hyaline spherical resting spores (Francis, 1981). Seeds partly or completely encrusted with oospores often appear dull white and have cracks in the seed coat.

Plants developing from oospore-encrusted seed are systemically infected, remain stunted, and die early (Francis, 1981). Symptoms in infected seedlings appear when plants are about 2 weeks old. Light-green areas appear at the base of the primary leaves and spread along the veins in a serrated or fan-like manner. Not all trifoliate leaves show symptoms. Infected leaves may appear mottled and grey-green with curled down edges.

Known hosts

P. manshurica is known to attack soybean (*Glycine max*) primarily. It is also known to attack *Glycine soja,* which is a wild host.

Known Distribution

CABI International (2004) list of countries where downy mildew caused by *P. manshurica* has been reported: **Asia:** China, Iran, Israel, Japan, Kazakhstan, Korea, Malaysia, Phillipines, Thailand, Taiwan, Turkey, and Vietnam. **Europe:** Bulgaria, Croatia, Czech Republic, Denmark, France, Germany, Hungary, Italy, Latvia, Moldova, Poland, Romania, Serbia and Montenegro, Slovakia, Sweden, Ukraine, and the United Kingdom. **Africa:** Ethiopia, South Africa, and Zimbabwe. **North America:** Bermuda, Canada, Mexico, and the U.S. **Central America:** Cuba and Puerto Rico. **South America:** Argentina, Bolivia, Brazil, and Colombia. **Oceania:** Australia and New Zealand.

Potential Distribution Within the US

The pathogen is known to be present in Arkansas, Florida, Illinois, Iowa, Louisiana, Mississippi, North Dakota, North Carolina, Ohio, Oklahoma, South Dakota, and Wisconsin, but is most likely present across the soybean growing region of the U.S.

Survey

No specific surveys methods are published for *P. manshurica*. Incidence or severity of downy mildew may be determined by walking a soybean field in a ‘W’, ‘O’ or any other pattern, stopping periodically and examining the soybean plants closely for the typical symptoms.

Symptoms characteristic of downy mildew include: pale green to yellow spots on leaves, which enlarge to give irregular lesions that turn grayish-brown to dark brown with a yellowish margin in the upper leaf surface, corresponding with the growth of grayish to pale-purplish tufts (diagnostic sign) of conidiophores (Fig. 3) on the lower leaf surface during humid conditions. Interior of pods encrusted with

a whitish mass of mycelium and oospores. Whitish mass of mycelium and oospores on the seed coat. Dwarfing of whole plant.

Key Diagnostics

Differentiation from Asian soybean rust

Downy mildew is easily distinguished from Asian soybean rust by the growth of grayish to pale-purplish tufts of sporangiophores and sporangia (Fig. 3) on the lower leaf surface during humid conditions, whereas, Asian soybean rust forms pustules with pores releasing a powdery brownish-red mass of uredionospores.

Phakopsora pachyrhizi

Scientific Name
Phakopsora pachyrhizi Syd. & P. Syd.

Synonyms:
Phakopsora calothea, P. erythrinae, P. sojae, P. vignae, Physopella pacyrizi, Uromyces sojae, Malupa sojae, Uredo erythrinae, Uredo sojae

Common Name(s)
Asian soybean rust, Asiatic soybean rust, soybean rust

Type of Pest
Fungus

Taxonomic Position
Phylum: Basidiomycota, **Class:** Urediniomycetes, **Order:** Uredinales, **Family:** Melampsoraceae

Reason for Inclusion in Manual

Pest Description
Currently, there are two rust fungi that cause soybean rust, *Phakopsora pachyrhizi and P. meibomiae*. *P. pachyrhizi*, Asiatic soybean rust, has emerged as a major constraint of soybean production in both the eastern hemisphere (Australia, China, India, Taiwan, and Thailand) and in the western hemisphere (Brazil, Columbia, Costa Rica, Puerto Rico, and Hawaii). Another species of rust, *P. meibomiae* has been endemic in portions of South America for many years, but is considered less of a threat because it is not as aggressive as the Asiatic soybean rust. Both rust species have the same type of lesions and urediniospore morphology and thus cannot be distinguished except by using molecular tools. Asiatic soybean rust currently has a localized distribution within the U.S. It was detected for the first time in North American in Louisiana in November 2004 and, soon after, in other southeastern states of the U.S. This pest description will focus on *P. pachyrhizi*.

A rust fungus may produce as many as five different spore stages in its life cycle (Table 1). Production of all five stages by *Phakopsora pachyrhizi*, the soybean

rust pathogen, is uncertain (Green, 1984). *P. pachyrhizi* is described from the uredinial and telial stages. Like all rust fungi, *P. pachyrhizi* is an obligate parasite that requires living host cells.

Table 1. The five possible spore stages of a rust fungus.

STAGE	DESCRIPTION
0	Spermagonia bearing spermatia (n) and receptive hyphae (n)
I	Aecia bearing aeciospores (n+n)
II	Uredinia (uredia) bearing urediniospores (uredospores) (n+n)
III	Telia bearing teliospores (n+n → 2n)
IV	Basidia bearing basidiospores (n)

Spermatia (stage 0) and aecia (stage I) are not known to exist (Green, 1984).
Uredinia (stage II) are amphigenous (growing all around), most hypophyllous (on the under surfaces of leaves), minute, scattered or in groups on discolored lesions. Subepidermal in origin, the uredinia are surrounded by paraphyses arising from peridioid pseudoparenchyma; in addition, the uredinia have hymenial paraphyses. Openings are through the central apertures (ostioles). In appearance, the uredinia are pulverulent (appearing as if powdered); in color, uredinia are yellowish-brown to pale cinnamon-brown (Ono et al., 1992). Paraphyses (Fig. 2) are cylindric to clavate, 25 to 50 μm x 6 to 14 μm, slightly to conspicuously thickened apically (~18 μm). The color of the paraphyses ranges from pale yellowish-brown to colorless (Ono et al., 1992).

Figure 1. Soybean rust lesion with circular ostiole and urediniospores. Photo courtesy of USDA-ARS

Figure 2. *P. pachyrhizi* urediniospores and paraphyses, which appear identical to those of *P. meibomiae.* Photo courtesy of Mary Palm, USDA-APHIS-PPQ.

Urediniospores are sessile, obovoid to broadly ellipsoid, 18 to 34 μm to 15 to 24 μm, and minutely and densely echinulate (spiny) (Figs. 2, 3). The walls are uniformly about 1 μm thick. The color of the urediniospores ranges from pale yellowish-brown to colorless. In number, germ pores are mostly 4 to 8 (mostly 6, rarely 2 or 10). In position, germ

pores are equatorial or scattered on the equatorial zone; on occasion, germ pores are scattered on or above the equatorial zone (One et al., 1992).

Telia (stage III) are hypophyllus, often intermixed with uredinia, pulvinate and crustose, chestnut-brown to chocolate-brown, subepidermal in origin, and 2- to 7-spore layered. The teliospores are one-celled, irregularly arranged, angularly subglobose, oblong to ellipsoid, and (10-)15 to 26 um x 6 to 12 um. The wall is uniformly about 1 um thick, sometimes slightly thickened (up to 3 um) apically in the uppermost spores, colorless to pale yellowish-brown (Ono et al., 1992).

In 1984, Green noted that 'no germination of teliospores had been reported'. However, in 1991, Saksirirat and Hoppe reported germination of teliospores.

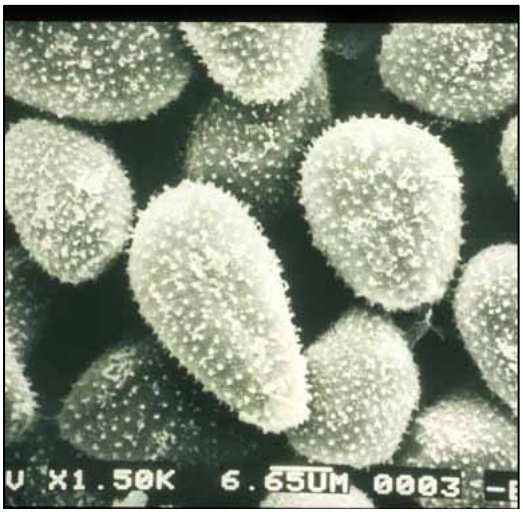


Figure 2. Scanning electron micrograph of soybean rust urediniospores showing spiny appearance. . Photo from the collection of Glen Hartman, USDA-ARS.

Biology and Ecology

Unlike many pathogens that must find stomata, wounds or some opening before they are able to penetrate the host, soybean rust urediniospores are able to penetrate directly through the leaf cuticle and epidermis, making infection easier and quicker. The incubation period for the fungus is about 7 days; while the latent period is about 9 to 10 days (Melching et al., 1979). In a histological study, Marchetti et al. (1975) found hyphae in soybean mesophyll 20 hours after inoculation with urediniospores of *P. pachyrhizi* and frequently observed direct penetration from appresoria formed at the end of short germ tubes, usually less than 20 um long.

P. pachyrhizi is believed to have a heteroecious life cycle. However, pycnial and aecial stages have not been found. In warmer regions, volunteer crops, supplementary legume crops, and wild species may harbor the fungus throughout the year or during seasons in which soybeans are not cultivated, and may serve as a primary infection source. In colder regions where above-ground parts of annual hosts senesce during winter, no source of new infections in the soybean-growing season has been identified.

Urediniospores of *P. pachyrhizi* germinated between 10 and 28.5 °C, with a broad optimum in the range of 15 to 25 °C. At optimum temperatures, urediniospores germinate in 1 to 1.5 hours. Maximal infection of 'Wayne' soybean occurred at 20 to 25 °C with 10 to 12 hours of dew and at 15 to 17.5 °C with 16 to 18 hours of dew. The minimal dew period for infection was 6 hours at

20 to 25 °C and 8 to 10 hours at 15 to 17.5 °C. Infection did not occur above 27.5 °C (Marchetti et al., 1976). The temperature-moisture requirements for the infection of soybeans by urediniospores of *P. pachyrhizi* would not preclude the establishment of the soybean rust fungus in all the major soybean growing areas of the U.S. (Marchetti et al., 1976).

Germinability and infectivity of urediniospores are reduced by exposure of the spores to dry and high temperature conditions prior to germination. Singh and Thapliyal (1977) reported that prior exposure of urediniospores to 35 °C for 6 hours prevented germination of an Indian isolate. Similarly, Kochman (1979) reported that germination of urediniospores on water agar at 21°C was significantly reduced by prior exposure of the spores to 28.5 to 42.5°C for 8 hours. According to Melching et al. (1989), urediniospores on unwetted soybean leaves progressively lost infectivity during sunny conditions, but exhibited enhanced infectivity after 1 or 2 days on dry foliage under cloudy conditions. After 8 days on dry foliage, no urediniospores were found to cause lesions following a 12 hour dew period at 18°C. Spores on leaves exposed to 4 or 6 hours of dew followed by drying for up to 4 days were able to infect when a 12 hour dew period was provided; however, they were less infectious than spores that had not been exposed to a brief initial wetting.

The formation of teliospores seems to be induced when infected plants are subjected to a temperature range below 20°C for at least 15 days. Yeh et al. (1981) reported that, on 20 soybean cultivars and nine other legume plants, teliospores were successfully induced when the inoculated plants were subjected to 12 hour photoperiods, 60 to 100% relative humidity (RH) and temperatures of 15 to 24°C. In the field, teliospores were produced only when the average daily temperature was below 20°C and the maximum temperature above 29°C. The authors further reported that telia and teliospores were formed on eight legume species when the infected hosts were inoculated and grown under a 12 hour photoperiod, at 60 to 100% RH, at a maximum day temperature of 24 ± 1°C and a minimum night temperature of 15 ± 1°C.

Dufresne et al. (1987) reported telial production in Taiwanese and Puerto Rican isolates. The two isolates were cultured on ‘Williams’ soybeans at two temperatures and three light intensities. The Taiwanese isolate produced telia after 21 and 30 days and the Puerto Rican isolate produced telia after 34 and 35 days at 10 and 15°C, respectively. At low light intensity (3.9 µE/m²/sec), the Taiwanese and Puerto Rican isolates produced telia after 29 and 33 days, respectively; at intermediate light intensity (5.3 µE/m²/sec) after 26 and 36 days, respectively; and at high light intensity (6.1 µE/m²/sec) after 22 and 34 days, respectively. The Taiwanese isolate produced larger lesions with a higher percentage of telia than the Puerto Rican isolate.

Saksirirat and Hoppe (1991) reported germination of teliospores. After treatment with 10 to 12 cycles of 24 hour wetting and 24 hour drying periods at room

temperature, 65 to 70% of teliospores germinated at 20°C under artificial illumination of 5000 lux at 12 hour light/dark intervals. Only 25% of teliospores germinated when the telia were treated with seven wetting and drying cycles. Higher germination rates were observed when telia were stored at 5°C for 5 to 6 months.

Pest Importance

Asian soybean rust is a serious disease of soybeans. Until recently this disease did not occur on soybean in the Western hemisphere, but it spread to South America in 2001 and was found for the first time in North America in November 2004. Soybean rust can be a devastating disease with yield losses up to 70 to 80% reported in some fields in Taiwan (Bonde et al., 1976). Plants that are heavily infested have fewer pods and smaller seeds that are of poor quality. In countries in which soybean rust is an established problem, losses range from 10 to 80 percent. The severity of losses varies depending on susceptibility of the soybean variety, time of the growing season in which the rust becomes established in the field, and weather conditions during the growing season.

Soybean rust spores can be carried long distances by wind currents. In 1998, spores were blown 1,350 miles down Africa from Uganda to Zimbabwe. Between 2001 and 2003, the disease spread more than 1,500 miles, from Paraguay to near the equator, infecting as much as 90% of Brazil's soybean acres on the way (APHIS, 2004). Although the exact source of the infection in the continental U.S. is unknown, a probable explanation is the spread of the disease from South American to the U.S. during the active hurricane season.

Unlike other rusts, *P. pachyrhizi* and *P. meibomiae* infect an unusually broad

Figure 4. Soybean infected with soybean rust in Parana State BS near Londrina, Brazil; From left to right unsprayed, sprayed once with a fungicide and sprayed twice with a fungicide. Photo courtesy of Steve Koenning, North Carolina State University.

range of plant species, which increases the importance of the pest. *P. pachyrhizi* naturally infects 31 species in 17 genera of legumes, and 60 species in 26 other genera have been infected under controlled conditions. Twenty-four plant species in 19 genera are hosts for both species.

It has been estimated that yield losses from *P. pachyrhizi* could exceed 10% in most of the U.S. and up to 50% in the Mississippi Delta and southeastern U.S. Currently, there is no resistance to soybean rust in any of the U.S. commercial soybean cultivars. Some fungicides are effective against *P. pachyrhizi* by slowing the spread of the pathogen enough so that normal seed set and pod fill can occur (Fig. 4) However, widespread fungicide applications on soybeans in the U.S. are not deemed cost effective. As a result this control option would be useful only for eradication on small acreages (Koenning et al., 2004).

Symptoms/Signs

The first symptom of soybean rust is chlorosis (Fig. 5A). Early symptoms of rust infection are found on leaves deep in the canopy, and look like tiny black specks scattered with mottled yellow areas (Fig. 5B). These yellow areas appear translucent if the affected leaves are held up to the sun. Asiatic soybean rust forms two types of lesions on leaves, tan and reddish brown. Lesions will contain one to three rust pustules, which are raised on the leaf surface. The lesions may have an angular appearance and be limited by leaf veins. Rust pustules may appear on cotyledons, leaves, petioles, stems, or pods, but are most likely to be observed as raised pustules on the under side of the leaf (abaxial) (Fig. 6). Soybean rust pustules are small (about the size of a pin head) and contain hundreds of spores. Spores are elliptical to obovoid in shape, colorless to yellowish or yellowish brown and minutely and densely spiny. Infected plants will senesce early and have smaller seeds with reduced yield (Koenning et al., 2004). For the rust to cause economic damage, first infections will probably have to occur before the R3 stage of soybean development.

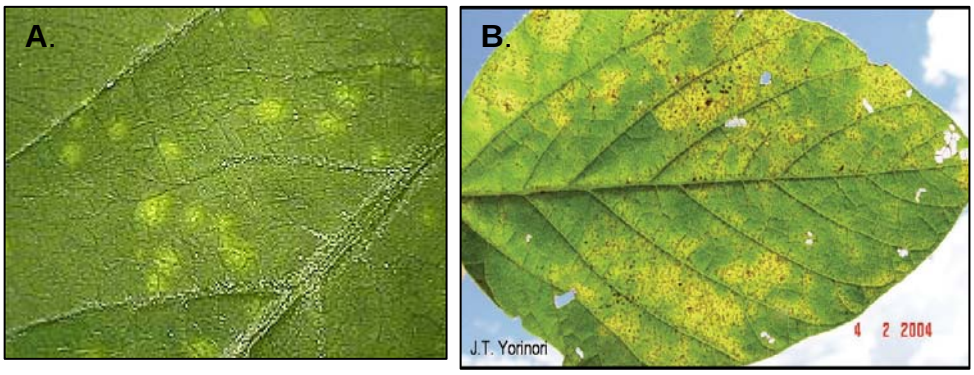


Figure 5. Early soybean rust infection symptoms on susceptible soybean. A) Chlorosis and B) black specks surrounded by mottled yellow areas. Photo courtesy of Glen Hartman and J.T. Yorinon.

On susceptible species/cultivars, infections result in small yellowish-brown or greyish-brown spots or lesions (TAN-type) (Fig. 7), which are delimited by vascular bundles. Several pustules ('pimple-like' structures) of urediniospores are formed on both adaxial and abaxial surfaces of the lesion, but more frequently on the abaxial surface. The lesions coalesce, become dark brown and are covered by buff or pale-brown spore masses as sporulation progresses. The tan lesions when mature, consist of small pustules with masses of tan colored urediniospores on the surface. Later in the season, the lesions become dark reddish-brown and crust-like; these are subepidemal telial clusters. When resistant species/cultivars are infected, minute, reddish-brown spots (RB-type) appear, on which only a few uredinial pustules are formed. Sporulation on RB-type lesions is much less than on TAN-type lesions.

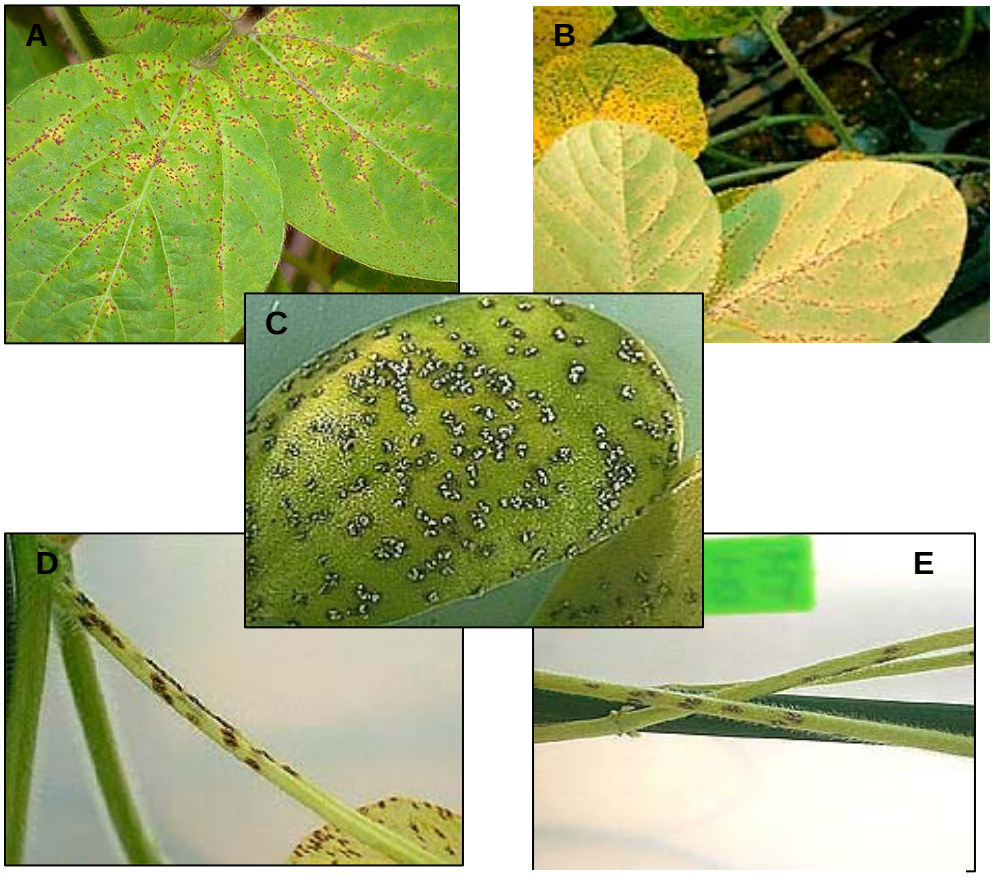


Figure 6. Soybean rust pustules caused by *Phakopsora pachyrhizi* (A) on the upper side of a soybean leaf, (B) on the under side of a soybean leaf, (C) on a soybean cotyledon, (D) on petioles, and (E) stems. Photo courtesy of Glen Hartman.

In soybean, *P. pachyrhizi* causes extensive necrosis of tissues in and around the penetration site. It may take weeks for productive uredia to appear within this necrotic zone. This is not typical of a majority of rust fungi. In soybean rust, the living hyphae must connect these uredia to food and water sources in living cells at distances up to perhaps 1 mm (Melching et al., 1979).

Known Hosts

Because of the confusion over the taxonomy of the pathogens causing soybean rust, *P. meibromiae* and *P. pachyrhizi*, the list of hosts of *P.pachyrhizi* may be incomplete; however, according to various recent references, a large number of legume species are host plants for *P. pachyrhizi. P. pachyrhizi* naturally infects 31 legume species in 17 different genera. *P. pachyrhizi* has been known to infect and sporulate in the field on 35 species in 18 genera of the Subfamily Papilionoideae in the Fabaceae. Among the naturally infected hosts, only *Crotalaria anagyroide*s, *Glycine max*, *Pachyrhizus erosus*, *Phaseolus lunatus*, and *Vigna unguiculata* serve as hosts of another soybean rust fungus, *Phakopsora meibomiae*, which occurs exclusively within the Americas.

Figure 7. Tan-type of soybean rust lesion. Photo courtesy of Glen Hartman.

Major hosts

Cajanus cajan (pigeon pea), *Glycine max* (soybean), *Lupinus* (lupine), *Pachyrhizus erosus* (yam bean), *Phaseolus* spp. (beans), *Pueraria montana* var. *lobata* (kudzu), and *Vigna unguiculata* (cowpea).

Minor hosts

Calpogonium mucunoides, *Erthrina subumbrans* (December tree), *Erythrina variegata* (Indian coral tree), *Kennedia prostrate*, *Kennedia rubicund*a, *Mucuna* (velvetbeans), *Pueraria phaseoloides* (tropical kudzu), *Vicia villosa* (winter vetch), and *Voandzeia subterranea* (bambara groundnut).

Because kudzu is a common weed in the southeastern U.S., it might serve as a continental source of inoculum.

Wild hosts

Glycine soja (wild soybean).

Known Distribution

Asia: Bangladesh, Cambodia, China, India, Indonesia, Japan, Korea, Laos, Malaysia, Myanmar, Nepal, Philippines, Russian Federation, Singapore, Sri Lanka, Thailand, Vietnam. **Africa:** Congo Democratic Republic, Ethiopia, Ghana, Mozambique, Nigeria, Sao Tome and Principe, Sierra Leone, South Africa, Sudan, Tanzania, Uganda, Zambia, Zimbabwe. **North America:** the U.S. **Caribbean:** United States Virgin Islands, Puerto Rico. **South America:** Argentina, Bolivia, Brazil, Paraguay, Uruguay, Venezuela. **Oceania:** Australia,

Cook Islands, Federated states of Micronesia, Guam, New Caledonia, Niue, Papua New Guinea, Tonga, Vanuata.
Asian soybean rust was first observed in Japan in 1902. Until recently the pathogen was distributed throughout Asia and Australia. It was reported from Hawaii in 1994. In the late 1990's Asian soybean rust was found in Africa and in 2001 was reported in South America. As of 2004, Asian soybean rust in the Americas was known from Argentina, Bolivia, Brazil, Paraguay, and Uruguay.

In November 2004, *P. pachyrhizi* was found for the first time in Louisiana and, soon thereafter, in other southeastern U.S. states. Many earlier reports of *Phakopsora pachyrhizi* in the Americas are erroneous. The reports of *P. pachyrhizi* prior to 1992 actually refer to *P. meibomiae*, a similar-looking rust that also occurs on soybeans and numerous other legumes. In a monograph of the genus Phakopsora Ono et al. (1992) discussed the morphological differences between *P. pachyrhizi* and *P. meibomiae*, although it is difficult to separate them based on morphology with certainty. A molecular test for differentiating these species was published by Frederick et al. (2002) and its use is essential for the accurate identification of these two species.

Potential Distribution Within the US

Soybean rust has a localized distribution within the United States. It was detected for the first time in North America in Louisiana in November 2004 (Scheider et al., 2005) and, soon after, in other southeastern states of the US (Hernandez, 2005). It was found on the alternate host kudzu (*Pueraria montana* var. *lobata*) in Florida in March 2005. It was also observed on Florida beggarweed (*Desmodium tortuosum*) in Georgia in November 2005. It has also been reported in Alabama, Arkansas, Georgia, Hawaii, Iowa, Mississippi, Missouri, North Carolina, and South Carolina.

Predictive models suggest that conditions in Georgia, South Carolina, Virginia, and North Carolina are favorable for development of an epidemic of soybean rust. The soybean rust pathogen is primarily tropical in distribution and would be able to survive over winter in only the most southern portions of the U.S. (southern Florida and Texas).

Survey

The disease is detected by inspecting the abaxial surface of leaves for uredinial pustules that are powdery and buff and pale brown. The disease is diagnosed both macroscopically by the characteristic symptoms and microscopically by abundantly paraphysate uredinia with pale-yellowish brown or almost colorless, echinulate uredinospores (CABI, 2004).

One of the challenges of identifying Asian soybean rust is that the early stage of the disease can look like other leaf diseases of soybean (see brown spot, bacterial blight, bacterial pustule, frogeye leaf spot, *Cercospora* leaf blight and downy mildew sections of this manual). In general, to check a field for rust: walk

through the entire field in a standard scouting pattern (e.g. a 'W'-shaped pattern), periodically stop and examine the soybean plants, look low and deep into the canopy of the plants, and closely examine the plants for mottled yellow leaves with 'tell-tale' pustules (pimple-like structures) on the underside. Areas in the field with distinct yellowing or browning of the leaves, or areas of dense canopy development, should be targeted in addition to the areas covered by the standard scouting pattern.

Key Diagnostics

P. pachyrhizi is considered an Australasian species of soybean rust and *P. meiborniae* is a new world species. *P. pachyrhizi* is the species currently causing damage in the Southern Hemisphere. Both have the same type of lesions and urediniospore morphology, and thus cannot be distinguished except by using molecular tools. Classical and real-time PCR techniques were developed by Frederick et al. (2002) to detect soybean rust and distinguish the soybean pathogens *Phakospora pachyrhizi* and *P. meiobomiae*.

Bacterial pustule caused by *Xanthomonas axonopodis* pv. *glycines* and bacterial blight caused by *Pseudomonas savastanoi* pv. *glcinea* produce spots similar to those formed by the soybean rust fungus on the discolored leaf lesions. However, the bacterial spots are at first water-soaked in appearance and later ooze out slimy bacterial masses instead of powdery spore masses in the rust (CABI, 2004). Bacterial pustule is also rare in commercial soybean varieties, since most if not all are resistant to this disease. A hand lens may aid in seeing the raised nature of the pustule. Also, placing leaves in a plastic bag with a moist paper towel for twenty four hours may cause the pustules to erupt, thus making identification easier.

Septoria glycines

Scientific Name
Teleomorph: *Mycosphaerella uspenskajae* Mashkina & Tomilin, Mikol.
Anamorph: *Septoria glycines* Hemmi, Trans. Sapporo

Common Name(s)
Brown spot of soybean, soybean brown spot, soybean leaf spot

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota, **Class:** Ascomycetes, **Order:** Mycosphaerellales, **Family:** Mycosphaerellaceae

Reason for Inclusion in Manual

Pest Description
According to Punithalingam and Holliday (1972), *Septoria glycines* forms an asexual fruiting body called a pycnidium in the dead tissues of old lesions. Pycnidia are mostly epiphyllous, immersed, yellow-brown to dark brown, subglobose, and 100 to 180 µm in diameter that become erumpent with ostioles (40 to 70 µm). The pycnidial cell wall is 2 to 4 layers thick and composed of yellow brown cells that are thickened on the outside. Pycnidia in leaf tissues are globose to conical globose, whereas those in stems are flattened (Sinclair and Hartman,

Figure 1. Conidia of *S. glycines* at 400 X. Photo courtesy of P. R. Sellers.

1999). The conidiophores arising from the cells lining the inside of the pycnidium are hyaline, obclavate to obpyriform, and 6 to 10 x 3 to 4 µm in size. Conidia (pycnidiospores) are hyaline, typically filiform, straight or curved (Fig. 1), guttulate, with basal end blunt, gradually tapering, rounded at apex, and 30 to 50 x 1.5 to 2 µm in size, with 1 to 4 septa noticeable at germination. Conidia readily germinate in water on the surface of leaves (Punithalingam and Holliday, 1972; Sinclair and Hartman, 1999).

Biology and Ecology

S. glycines overwinters in soybean residues left on the soil surface and in infected seeds. From these primary inoculum sources, the pathogen is spread to young plants by wind and splashing rain. Infection occurs on lower leaves as early as the V2 growth stage. The pathogen enters the leaves through stomata and grows intercellularly, killing cells next to the hyphae. It also penetrates pods through stomata and seeds via the placenta and funiculus tissue. Inoculum on lesions in infected cotyledons and in unifoliate leaves serve as sources of secondary inoculum. Under favorable weather conditions (warm and wet), the disease progresses throughout the plant and leads to premature senescence, even at low disease severity (Schuh and Adamowicz, 1993). Late in the growing season, infected leaves may turn rusty brown or yellow and drop prematurely. The spread of the disease is usually halted during hot, dry weather, but it can redevelop again before soybean plants mature (Sinclair and Hartman, 1999).

Severity of brown spot is strongly influenced by temperature and the duration of leaf wetness period. In general, disease severity increases with increasing leaf wetness periods from 6 to 36 hours. The optimum temperature for brown spot development is 25 ºC, but the disease can develop from 15 ºC to 30 ºC (Sinclair and Hartman, 1999).

Figure 2. Symptoms of brown spot. Photo courtesy of X. B. Yang.

Cultivars vary in their susceptibility to *S. glycines* (Sinclair and Hartman, 1999). Cultivars with partial or rate-reducing resistance can be used to control the pathogen. Since brown spot is more severe in continuously cropped soybean fields, crop rotation is useful. If economically feasible, applications of foliar fungicides from bloom to pod fill may reduce the severity of disease (Sinclair and Hartman, 1999). In fields with very high levels of brown spot, plowing of straw may promote a rapid decay of these residues.

Pest Importance

Brown spot rarely causes economic yield reductions. Its primary effect is premature defoliation and reduced seed size. However, some yield loss may occur during extremely wet growing seasons. These yield losses occur mostly in high yielding environments and are related to timing and rate of defoliation. If 25 to 50% of the canopy is defoliated prematurely, yield losses in the range of 8 to 15% may occur. Brown spot severity at the R6 growth stage is predictive of the potential yield loss.

Symptoms

Brown spot of soybean is primarily an early season foliar disease that typically occurs on the lower leaves. Cotyledons, primary leaves and lower trifoliolate leaves show brown to reddish brown pinpoint spots that can be up to 4 mm in diameter (Fig. 2). Lesions appear on both upper and lower surfaces. Lesions may coalesce and become irregularly necrotic blotches (Fig. 3). Tiny black points (pycnidia) develop in the center of older lesions. Severe infection can cause leaves to yellow and drop early, especially those in the lower canopy. Numerous irregular light brown lesions form on trifoliate leaves. These lesions gradually darken until they become chocolate brown to blackish brown. On the main stem, branches, petioles, and pods, lesions are brown with indefinite margins. These lesions range from small specks to areas of several square centimeters and usually have pycnidia. However, symptoms are not sufficiently distinct from those of other soybean diseases to be diagnostic.

Figure. 3. Symptoms of Septoria brown spot. Note the large rusty brown lesions. Photo Courtesy of P. Lipps.

Known Hosts

Main host for *S. glycines* is soybean (*Glycine max*).

Lee and Hartman (1996) reported that the following legume species developed symptoms of brown spot after inoculation in the field and/or greenhouse with *S. glycines*: *Glycine arenaria*, *G. argyrea*, *G. curvata*, *G. soja*, *G. tabacina*, *G. tomentella*, *Lablab purpureus*, *Lens culinaris*, *Lupinus albus*, *L. mutabilis*, *Medicago sativa*, *Onobrichis viciifolia*, *Phaseolus coccineus*, *P. lunatus*, *P. vulgaris*, *Pisum sativum*, *Trigonella foenum-graecum*, *Vicia faba*, *V. hirsuta*, *V.*

sativa, *Vigna angularis*, *V. mungo*, *V. sesquipedalis*, and *V. unguiculata*. Also, *Amphicarpa bracteata* and *Abutilon theophrasti* (velvetleaf) are known to be hosts of *S. glycines*.

Known Distribution

CABI International (2004) lists the following countries where brown spot can be found. **Asia:** China, India, Japan, Korea, and Nepal. **Europe:** Germany, Italy, Poland, Romania, Serbia and Montenegro, and the USSR. **Africa:** Zimbabwe. **North America:** Canada and the U.S. **South America:** Bolivia, Brazil, and Colombia.

Potential Distribution Within the US

Brown spot was first reported in North Carolina in 1923, and now is widespread in Arkansas, Delaware, Florida, Illinois, Indiana, Iowa, Maryland, Minnesota, Missouri, North Carolina, South Dakota, and Wisconsin.

Survey

No specific survey methods have been published for brown spot. Since brown spot is usually the first disease to appear on young plants, surveys may start at V2 stage. Later surveys may be carried in coincidence with surveys aimed to detect other soybean foliar diseases such as Asian soybean rust. During surveys, avoid checking only the perimeter of fields. Walk fields in a 'W' shaped pattern or other survey type pattern. Check at least 6 to 10 locations within each field. Check areas where moisture conditions are favorable for disease development, including low-lying areas, along roads, and near bodies of water.

Look for angular to somewhat circular, dark reddish-brown spots on both upper and lower leaf surfaces (Fig. 2). Adjacent lesions frequently merge to form irregularly shaped blotches, and leaves become rusty brown (Fig. 3). Locate the black dots (pycnidia) in center of mature spots. Crush tissue with pycnidia in a small drop of water and identify the pycnidiospores (conidia) of *S. glycines* under the microscope (Fig. 1).

Key Diagnostics

Differentiation from Asian soybean rust

S. glycines can be confused with *Phakopsora pachyrhizi*. Two distinct types of brown spot lesions have been described on soybeans. The most common type is an angular reddish brown lesion surrounded by a chlorotic halo and is associated with plants grown from yellow seeds. The other type is an angular dark brown lesion without the surrounding chlorosis and is associated with plants grown from green seeds (Lim, 1979). In contrast, lesions of Asian soybean rust are initially yellow flecks or tan to brown or reddish-brown pinpoint spots on the upper leaf surface. As they get older, these lesions develop pale brown pustules on the undersides of leaves from which masses of uredionospores are released.

Viral Diseases

Bean Common Mosaic Virus (BCMV)

Scientific Name
Bean Common Mosaic Potyvirus

Common Name(s)
Bean common mosaic virus –serotype B, bean mosaic virus, bean virus 1, bean western mosaic virus, common bean mosaic virus, mungbean mosaic virus, Phaseolus virus 1

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Many strains of the Bean common mosaic virus (BCMV) have been distinguished. Those once grouped as serotype A are now considered isolates of a separate potyvirus species, bean common necrosis virus, and several viruses once considered distinct, have now been shown to be strains of the virus. The latter include: akuki bean mosaic virus, blackeye cowpea mosaic virus, cowpea (aphid-borne) mosaic virus, cowpea (blackeye) mosaic virus, cowpea vein-banding mosaic virus, peanut blotch virus, peanut stripe virus, and some isolates from soybean.

BCMV has flexuous filamentous particles, 720 to 770 nm long and 12 to 15 nm wide. The particles are composed of 95% protein, usually of one main polypeptide species of 32 to 35 kDa. A component of 29 kDa may also be found in virus preparations, which have undergone limited proteolysis. The remaining 5% of the particle is made up of single-stranded RNA.

The particles form a single sedimenting and buoyant density component and have a sedimentation coeffecient value of 154 to 158 S (measured for US1 and US5 strains) and a buoyant density in caesium chloride of 1.31 to 1.32 g/cm^3.

The stability of virus in sap is dependent on the strain and source of virus, and on test conditions. The thermal inactivation point ranges from 50 to 60°C, the dilution end point is between 10^{-3} and 10^{-4}, and the virus retains its infectivity in sap for 1 to 4 days at room temperature (CABI, 2004).

Plant cells infected with BCMV develop characteristic cytoplasmic inclusion bodies of the pinwheel associated scroll type described by Edwardson and Christie (1978).

Biology and Ecology

Vectors provide a means of secondary spread of BCMV within a crop or primary infection of a healthy crop. BCMV can be transmitted in the non-persistent manner by several aphid species, which do not normally colonize *P. vulgaris* but transmit BCMV as winged migrants, especially *Acyrthosiphon pisum*, *Aphis fabae* and *Myzus persicae* (Kennedy et al., 1962; Zettler and Wilkinson, 1966).

Several other aphid species transmit BCMV including *Aphis gossypii, A. medicaginis*, *A. rumicis*, *Hayhurstia atriplicis*, *Uroleucon ambrosiae*, *Macrosiphum euphorbiae* and *Acyrthosiphon pisum* (Zaumeyer and Thomas, 1957).

The efficacy of BCMV transmission is determined by the pre- and post-feeding behavior of *Myzus persicae* (Zettler and Wilkinson, 1966). Zettler (1969) demonstrated that the availability of the virus to aphids was dependent on symptom expression. Chlorotic areas were better sources of virus for transmission; leaves formed soon after inoculation were better sources of virus than older leaves.

BCMV is seedborne on a range of legumes. Klein et al. (1988) found BCMV in accessions of *Phaseolus vulgaris*, *P. acutifolius*, *P. aborigineus* and *P. angustifolius* seeds in a survey of the USDA *Phaseolus* germplasm collection. BCMV appeared to be a serious problem in *P. vulgaris* accessions; approximately 60% of the 207 tested were contaminated with incidences of infection of up to 70% being found in individual accessions. Other hosts on which seedborne BCMV has been detected include *Clitoria ternatea* (Lima et al., 1993), cowpeas (Patil and Gupta, 1992), mung beans, *P. coccineus* (Chamberlain, 1939), *P. acutifolium* var. *lactifolius* (*Phaseolus acutifolius*) (Provvidenti and Cobb, 1975), *Macroptilium lathyroides* (Provvidenti and Bravermen, 1976) and *Vigna mungo* (Agarwal et al., 1979a, b).

Early work reported BCMV as being transmitted by pollen (Reddick, 1931). More recently, electron microscopy of leaf extracts and pollen grains of BCMV-infected bean plants revealed filiform particles 750 µm long, which were absent from healthy plants (Omar et al., 1978a). In another report (Omar et al., 1978b), BCMV was detected in sepals, petals, stamens and pistils; infectivity was highest in parts of the pod and embryo of immature bean seeds. The virus was not

present in cotyledons and disappeared from the seedcoats of mature dried seeds. This study also showed that plants infected at an early stage produced up to 57% of seeds carrying the virus. Hagita et al. (1975) showed that when the primary leaves were inoculated, seed transmission was 89% compared with 40%, 5% and 1%, respectively, following inoculation of the first, second, and third trifoliate leaves. No infection occurred when inoculation was delayed until 7 to 10 days after flowering. The importance of time of plant infection is also indicated by transmission of the pathogen by seeds: maximum seed transmission was achieved when plants were inoculated at the primary leaf stage, whereas few cultivars transmitted the virus in seed when plants were inoculated 30 days after sowing (Morales and Castano, 1987).

The virus is located mainly in the embryo (Provvidenti and Cobb, 1975) but also exists in other seed parts (Ekpo and Saettler, 1974; Raizada et al., 1990). Monoclonal antibody ELISA of 1350 seeds indicated erratic virus distribution in the individual seed parts including seedcoat, testa and cotyledon (Klein et al., 1992).

BCMV has considerable longevity in bean seed; particles have been reported to remain infective in bean seed for up to 30 years (Zaumeyer and Thomas, 1957). Seed transmission levels of 3 to 4% were found in seed lots even after storage for more than 6 years at 2 to 4°C (Jeyanandarajah and Brunt, 1993). BCMV had no effect on bean seed germination, but specific weight and seed index were significantly reduced by infection (Omar et al., 1978c).

Pest Importance

BCMV can be found throughout the world, wherever beans are grown. The virus was considered to be of minor importance in the U.S. after the introduction of seed certification and dominant resistance (Provvidenti, 1990). However, severe epidemics of necrotic strains have occurred in recent years in the north-western area of the U.S. In Michigan, only the NY15 and type strains were present until 1982, when a severe necrotic strain was found (Kelly et al., 1982). Further investigation revealed that this strain resembled the NL3 strain (Kelly et al., 1984).

BCMV is economically important throughout Africa, Europe, North America and Latin America. Infection levels may reach 100% and estimated yield losses range from 35 to 98% (Galvez, 1980). Yield reductions in bean crops due to BCMV ranged from 53 to 68% in Oregon depending on disease severity (Hampton, 1975).

Severe outbreaks of BCMV occurred in Morocco in 1972 and 1974, in which 50% of all bean plantings showed BCMV symptoms from a seedborne infection spread by aphids (Lockhart and Fischer, 1974); yield losses were estimated at 50% and 34% infection of harvested seed.

Symptoms/Signs

BCMV causes two types of symptoms: common mosaic and common mosaic necrosis (black root) in *Phaseolus vulgaris*. The occurrence of either type of symptom depends on the particular virus present and whether or not the bean cultivar possesses the dominant I resistance gene. If the cultivar has the dominant I resistance gene, it is resistant to strains of the BCMV, but hypersensitive to strains of the Bean common mosaic necrosis virus (BCMNV).

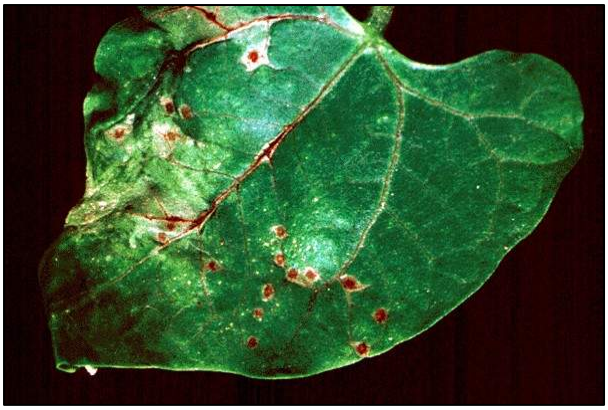


Figure 1. Blistering and leaf roll caused by BCMV. Photo courtesy of CABI, 2004.

Symptoms associated with common mosaic include leaf rolling or blistering (Fig. 1), light and dark-green patches on the leaf (green mosaic) (Fig. 2), chlorotic vein banding, yellow mosaic and growth reduction (CABI, 2004). Mottling and malformation of the primary leaves is an indication that the primary infection occurred through seed (Galvez, 1980). Cultivars that develop common mosaic may have distinct chlorotic or necrotic local lesions, which are not associated with the vascular system.

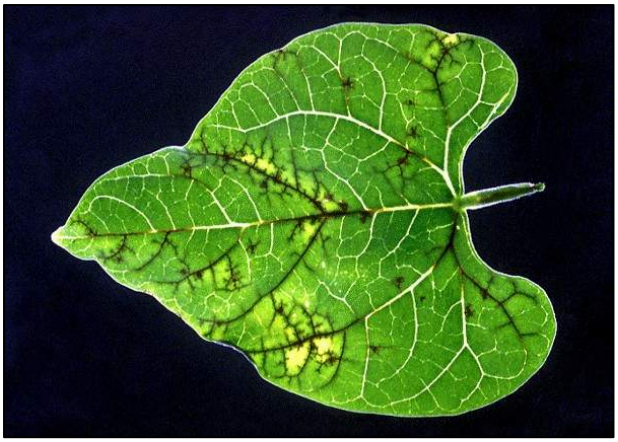


Figure 2. Light and dark-green patches on the leaf, chlorotic vein banding, yellow mosaic caused by BCMV. Photo courtesy of CABI, 2004.

Systemically infected plants may have smaller and fewer pods, and infected pods may sometimes be covered with small, dark-green spots and mature later than uninfected pods (Zaumeyer and Goth, 1964; Zaumeyer and Thomas, 1957).

Black root is characterized by local necrotic lesions, which extend into the veins causing systemic necrosis in the vascular system; this symptom only occurs in cultivars possessing the dominant resistance gene I. This necrosis can extend into the roots, stem and meristem and may result in plant death if the plant is infected at an early stage. If infected at a later stage, the plant may survive but parts may die. The pods may become discolored and unmarketable (Drijfhout, 1978; Morales and Bos, 1988).

Known Hosts

Natural hosts of BCMV are mainly restricted to *Phaseolus* spp., especially *P. vulgaris*.
BCMV has been isolated naturally from other leguminous species including: *Cajanus cajan* (pigeon pea), *Glycine max* (soybean), *Cassia tora*, *Chenopodium amaranticolor*, *Cicer arietinum*, *Crotalaria juncea*, *Crotalaria spectabilis*, *Crotalaria striata, Cyamopsis tetragonoloba*, *Gomphrena globosa*, *Lens culinaris*, *Vigna unguiculat*a, *V. radiata*, *Vicia sativa*, *Vicia villosa*, *Rhynchosia minima*, *Lupinus albus*, *Lupinus angustifolius*, *Lupinus luteus*, *Melilotus albus*, *Nicotiana benthamiana*, *Nicotiana clevelandii*, *Sesbania exaltata*, *Tetragonia tetragonioides*, *Trifolium incarnatum,* and *Trifolium subterraneum*,

Known Distribution

BCMV can be found wherever *Phaseolus* beans are grown. This includes many temperate, subtropical and tropical regions of the world. Countries with the virus present include: **Asia:** China, Indian Indonesia, Iran, Iraq, Israel, Japan, Kazakhstan, Korea, Lebanon, Saudi Arabia, Thailand, Turkey, Yemen; **Europe:** Belgium, Bulgaria, Czech Republic, Finland, France, Germany, Greece, Hungary, Italy, Lithuania, Netherlands, Norway, Poland, Portugal, Romania, Russian Federation, Spain, Sweden, Ukraine, United Kingdom; **Africa:** Burundi, Congo Democratic Republic, Egypt, Ethiopia, Kenya, Lesotho, Malawi, Mauritius, Morocco, Mozambique, Rwanda, Sierra Leone, South Africa, Sudan, Swaziland, Tanzania, Togo, Uganda, Zimbabwe, Bermuda; **North America:** Canada, Mexico, U.S.; **Central America:** Costa Rica, Cuba, Dominican Republic, El Salvador, Guatemala, Haiti, Jamaica, Nicaragua, Puerto Rico, Saint Vincent and the Grenadines, Trinidad and Tobago, **South America:** Argentina, Brazil, Chile, Ecuador, Colombia, Guyana, Peru, Venezuela; **Oceania:** Australia, Fiji, and New Zealand (CABI, 2004).

Potential Distribution Within the US

The virus is known to be present in California, Idaho, Michigan, New York, and Washington. However, the virus is thought to be more widespread within the U.S. (CABI, 2004).

Survey

There are no specific survey methodologies available for BCMV. Leaves should be inspected for signs of mosaic, chlorosis, necrosis or distortion (CABI, 2004).

Key Diagnostics

There are five diagnostic hosts for BCMV:

Chenopodium quinoa: faint chlorotic lesions developing into green rings; not systemic.

Macroptilium lathyroides: necrotic local lesions; systemic necrosis

Phaseolus vulgaris (cvx. Dubbele Witte, Stingless Green Refugee): green vein-banding, malformed leaves.

Pisum sativum: symptomless

Vicia faba: symptomless

Eleven host genotypes of *P. vulgaris* can be used to differentiate BCMV isolates into 10 pathotypes on the basis of systemic infection (Drijfhout, 1978). Isolates can be assigned to pathotypes according to the reaction patterns of the differential host cultivars (Spence and Walkey, 1995).

A number of monoclonal antibodies and polyclonal antisera can be used in ELISA to aid the identification of BCMV isolates (Spence and Walkey, 1995). Species-specific monoclonal antibodies are available for the identification and differentiation of serotype A and serotype B isolates (Mink et al., 1994). Monoclonal antibodies possessing three epitopes located on the coat protein amino terminus of viruses of the BCMV group have been found to differentiate some group members (Mink et al., 1999).

Using partial nucleotide sequences of selected isolates of BCMV and BCMNV, a reverse transcription, polymerase chain reaction (RT-PCR), in combination with restriction endonuclease analyses, has been developed for the molecular detection of BCMV, BCMNV and some viral pathogroups (PG). Specific detection of the two viruses was accomplished by constructing two virus-specific primer pairs that amplified a PCR product specific for each virus. By application of RT-PCR, four BCMV-PG-V isolates were differentiated from isolates of BCMV pathogroups I, II, IV and VII. Distinction of two BCMNV pathogroups (PG-III and PG-VI) was achieved by restriction enzyme XbaI digestion of BCMNV PCR products. However, no combination of tested restriction enzymes distinguished all five recognized BCMV pathogroups. A primer pair Dts/Uny15 proved to be specific for BCMV pathogroup PG-V. It is concluded that by a combination of RT-PCR and restriction enzyme analyses, it is possible to differentiate both viruses, two pathogroups of BCMNV and one pathogroup of BCMV from the others (Xu and Hampton, 1996).

Bean Golden Mosaic Virus (BGMV)

Scientific Name
Bean golden mosaic bigeminivirus

Common Name(s)
Bean golden mosaic geminivirus, bean golden mosaic virus (type 1) Brazil bean golden yellow mosaic virus (type 2) Caribbean, mosaico dorado, bean calico mosaic bigeminivirus

Type of Pest
Plant pathogenic virus

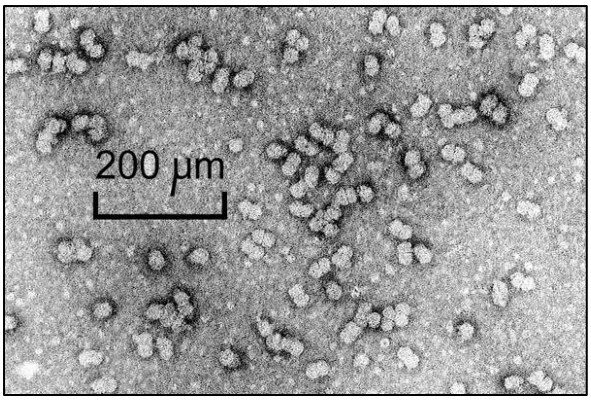


Figure 1. Geminate particles of BGMV. Photo courtesy of University of Florida, IFAS, TREC (CABI, 2004).

Reason for Inclusion in Manual

Pest Description
Bean golden mosaic virus (BGMV) particles are geminate (forming by pairing of two spherical particles) (Fig. 1); not enveloped; paired particles are 18 to 20 nm in diameter and 30 nm in length; profiles angular; capsomere arrangement not readily seen. Leaf sap contains few particles. In electron microscopy, aldehyde is necessary for fixation unless phosphotungstic acid (PTA) is used at pH 4 (CABI, 2004).

Biology and Ecology
Bean golden mosaic of common bean (*Phaseolus vulgaris)* was first found in Brazil in 1961 by Costa (1965). He also showed that the causal agent was transmitted by the whitefly *Bemisia tabaci*. BGMV incidence has steadily increased in the Caribbean islands, Cuba, Dominican Republic, Jamaica, Puerto Rico, Central America and southern Mexico where crops such as potato, cucurbits and tomato that attract whiteflies (*B. tabaci* and *B. argentifolii*) are grown near beans (Goodman et al., 1977; Galvez and Morales, 1989; Morales, 1994).

The whitefly *Bemisia argentifolii* was first described by Bellows et al. (1994). Yuki et al. (1998) showed that *B. argentifolii* was an effective vector of BGMV and that there was no marked difference in efficiency of transmission by *B. argentifolii* and

B. tabaci.

The whiteflies *B. tabaci* and *B. argentifolii* transmit BGMV. Adult *B. tabaci* and *B. argentifolii* may acquire the virus from infected plants in as little as 6 minutes, but efficient transmission requires longer feeding periods. Whiteflies can transmit the virus for periods ranging from a few days to several weeks and probably through the last molt. Transmission efficiency of an individual insect is often intermittent and erratic. Male and female whiteflies transmit with equal efficiency. No evidence has been found that BGMV can be transmitted through the ovaries (Haber et al., 1981).

In comparative transmission studies by males and females of *B. tabaci*, females were found to be more efficient than males in transmitting the virus to *Phaseolus acutifolius, P. polystachis* and *P. vulgaris*. However, females were less efficient in transmitting the virus to *P. longipedunculatus* and *P. lunatus*. BGMV is not reported to be seedborne (CABI, 2004).

Pest Importance

BGMV is a devastating disease of *Phaseolus vulgaris* in tropical America. Early or late infection by BGMV under field conditions in Brazil reduced the number of pods per plant, seeds per pod, yield, seed weight and seed germination. The reduction was greater with early infection (73%) than with late infection (43%) (Almeida et al., 1984). In trials with bean (*Phaseolus vulgaris*) cv. carioca 6C2, infection with BGMV reduced the yield of consumable beans by 64%, seed yield by 71%, weight of 100 seeds by 36.8%, seed germination by 4.8% and emergence by 5.3% (Menten et al., 1980).

BGMV was found in bean plants in all provinces in Cuba, with incidence reaching 100% in some areas. No pods were formed on infected plants (Blanco-Sanchez and Bencomo, 1981). Losses in bean crops caused by BGMV, transmitted by *B. tabaci,* can reach 100% in Sao Paulo, Brazil (Menten and Roston, 1980). Early spring losses in bean crops due to BGMV, transmitted by *B. argentifolii*, can reach 100% in southern Florida (McMillan, 1994). Bean production declined steadily between 1992 and 1994 with the incidence of BGMV in

Figure 2. Symptoms of BGMV on lima bean. Photo courtesy of Rob Williams (CABI, 2004).

bush beans ranging from as high as 100% to as low as 5%, and in pole beans BGMV infection may be as high as 100% to as low as 1% (McMillan et al., 1994).

Symptoms/Signs

Symptoms of BGMV in bean seedlings begin with fine, vein-limited lines of bright yellow chlorosis (Fig. 2), usually on the first trifoliate leaves emerging after inoculation. These fine chlorotic lines typically appear first on only half of the leaf near the leaf tips. Within 3 to 5 days, veinal chlorosis spreads to cover one-third or more of the leaf area, giving a characteristic net-like appearance, with bright yellow veins contrasting with the dark green interveinal areas. Veinal chlorosis later expands into a bright golden mosaic that has a striking appearance in the field. Younger trifoliate leaves emerging after the first leaves develop symptoms frequently become curled downwards within a week of the initial appearance of reticulate chlorosis (Fig. 3). Curling leaves fail to expand properly and their surfaces become stiff and leathery. Leaves with chlorosis and distortion may become necrotic. Seed setting is very poor if plants are infected at a very early stage. Bean pods are usually curled when plants are infected with BGMV at the pinbean stage. There are minor differences in symptomatology among bean cultivars (Haber et al., 1981; Faria et al., 1994a).

Figure 3. BGMV infected snap bean. Note: downward curling leaves, veinal chlorosis, and stunting. Photo courtesy of University of Florida, IFAS, TREC (CABI, 2004).

Known Hosts

Bean golden mosaic, caused by BGMV, occurs in most tropical and subtropical areas of the New World where beans are grown.

Major hosts

Glycine max (soybean), *Phaseolus acutifolius* (tepary bean), *Phaseolus lathyroides* (Phasey bean), *Phaseolus lunatus* (lima bean), and *Phaseolus vulgaris* (common bean)

Minor hosts

Cajanus cajan (pigeon pea), *Calopogonium mucunoides*, *Nicotiana tabacum* (tobacco), *Pachyrhizus erosus* (yam bean), *Phaseolus coccineus* (runner bean), *Vigna angularis* (adzuki bean), and *Vigna mungo* (black gram)

Wild hosts
Euphorbia spp. (spurges) and *Phyllanthus* spp.

Known Distribution
BGMV is present in Nigeria, Mexico, the U.S., Costa Rica, Cuba, Dominican Republic, El Salvador, Guatemala, Jamaica, Nicaragua, Panama, Puerto Rico, Argentina, Brazil, Colombia, and Venezeula (CABI, 2004).

Potential Distribution Within the US
The virus is currently present in Florida (Blair et al., 1995).

Survey
Visual examination of the symptoms of BGMV on infected plants and pods is not conclusive evidence that infection is present.

Key Diagnostics
Inoculation of the first, second and third trifoliate leaves of six bean cultivars revealed that differentiation of the cultivars by symptoms and by virus concentration used were best with inoculation of the first leaf. Mechanical transmissibility of BGMV isolates depends on their geographic origin. Isolates from Central America, the Caribbean and Colombia can be mechanically transmitted, whereas isolates from Argentina and Brazil cannot (Fazio, 1985).

When BGMV-infected phloem cells are stained with azure-A, the inclusions present are large, blue-violet and unusually shaped; hexagonally packed crystalline arrays or loose aggregates of virion-like particles in the nucleus. In addition, there are other changes such as the occurrence of characteristic rings in nuclei, after nucleoli undergo hypertrophy (Christie et al., 1986; Brunt et al., 1996).

Extracts from healthy and infected bean plants had higher cytokinin levels in stem and leaf extracts (Fazio, 1981). Euphorbia mosaic virus has a dilution end point of 1:1000, a thermal inactivation point of 55 to 60°C and longevity of 24 h in vitro. BGMV had corresponding values of 1:100, 50°C and up to 4 weeks at 8°C (Bird et al., 1977).

A greenhouse inoculation method for BGMV using viruliferous whiteflies (*Bemisia tabaci*) was developed, which ensured that bean plants are inoculated at the same stage of development with a uniform amount of inoculum (Adames Mora et al., 1996). In the greenhouse, the reaction of bean plants to BGMV could be determined within 30 days after planting, whereas field evaluations require up to 65 days. In addition, BGMV symptoms began to appear between 5 and 8 days after inoculation (DAI).

Blair et al. (1995) found that bean plants with bright golden mosaic symptoms tested positive for geminivirus infection when extracts were probed (dot blots)

with A component DNA from a geminivirus infection in *Macroptilium lathyroides* or from the recently identified tomato mottle geminivirus, both from Florida. The bean samples did not react with probes prepared to the B components for either of these viruses. Hybridization probes prepared to A and B components of BGMV-H gave strong reactions with extracts from beans infected with BGMV isolates from Guatemala and from the Dominican Republic.

BGMV DNA in inoculated *Phaseolus vulgaris* plants was detected by dot blot hybridization. BGMV-specific DNA replicated better at 30°C than at 15 or 22°C. Only the first trifoliate leaves of plants inoculated on primary leaves and incubated at 30°C developed symptoms. The maximum BGMV-specific DNA content was detected 9 days after inoculation at 30°C. BGMV double-stranded DNA was detected 6 to 9 days after inoculation (Aozaki et al., 1989).

Cowpea mild mottle virus (CPMMV)

Scientific Name
Cowpea mild mottle carlavirus

Common Name(s)
Bean angular mosaic virus, eggplant mild mottle virus, groundnut crinkle virus, groundnut Ngomeni mottle virus, psophocarpus necrotic mosaic virus, tomato pale chlorosis virus, voandzeia mosaic virus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Cowpea mild mottle virus (CPMMV) has straight or slightly flexuous filamentous particles (Fig. 1), mostly measuring 650 x 12 nm, which sometimes have a loosely coiled external helix of unknown composition. It has physico-chemical properties typical of carlaviruses (Brunt and Kenten, 1973); the structure of the 3' terminus of its genomic RNA is also similar to that of carlaviruses. However, unlike definite aphid-borne carlaviruses, it can be transmitted by whiteflies (e.g. *Bemisia tabaci*) and induces brush-like or falcate inclusions (Fig. 1) within infected plants that are probably composed mostly of aggregated virus particles.

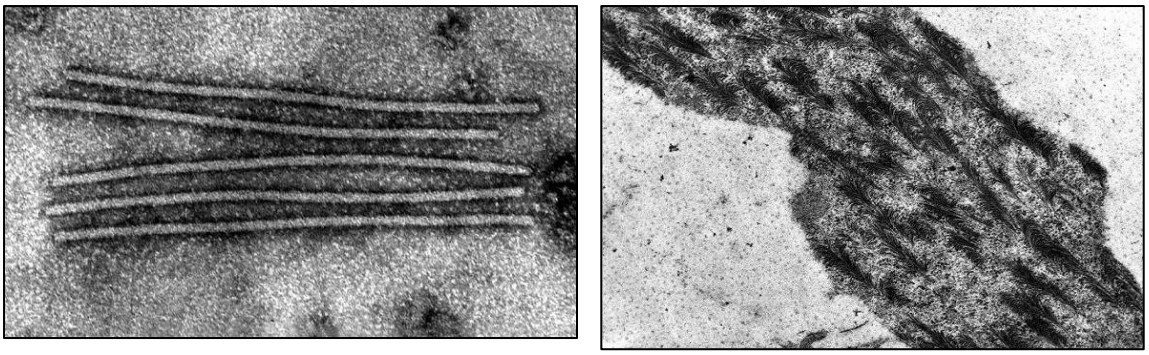


Figure 1. Transmission electron micrograph of CPMMV particles (left) and brush-like inclusions (right). Photo courtesy of Rothamsted Experimental Station (CABI, 2004).

It is, therefore, considered to be a tentative species of the carlavirus genus (Brunt, 1995).

Biology and Ecology

Bemisia tabaci, a whitefly, was first reported to be the natural vector of CPMMV by Iwaki et al. (1982). Laboratory transmissibility of the virus by this whitefly species has since been confirmed in Israel, Brazil, India, Nigeria, and Indonesia (CABI, 2004).

The virus was originally considered to be transmitted in the semi-persistent manner (Iwaki et al., 1982; Anno-Nyako, 1986). However, there is now cogent evidence that it is transmitted in the non-persistent manner (Costa et al., 1983; Muniyappa and Reddy, 1983).

The detailed epidemiology of CPMMV has yet to be investigated. Nevertheless, viruliferous whiteflies undoubtedly effect the transmission of virus from infected to healthy plants. Natural infection of perennial weed species has been reported in Kenya, Nigeria and India, and these are probably primary sources of infection for both tomatoes and leguminous crops. Similarly, when seed transmission occurs in leguminous crops, it may provide primary foci of infection for spread within a crop and transmission to adjacent tomato crops.

Reports of the seed transmissibility of CPMMV are contradictory. The original Ghanaian isolate, obtained from a seed-infected cowpea seedling, was subsequently shown to be seed-transmitted in cowpeas, soybeans, and French beans (*Phaseolus vulgaris*) (Brunt and Kenten, 1973). The virus was later reported to be seedborne to a level of 1 to 3% in cowpeas in India (Nain et al., 1994), to 0.9% in soybeans in Thailand (Iwaki et al., 1982), to 0.05 to 1.66% in 25 soybean cultivars in India, to unreported levels in soybeans in the Ivory Coast (Fauquet and Thouvenel, 1987), to unstated levels in cowpeas in India (Mali et al., 1989), and to 6 to 21% in bambara groundnuts (*Vigna subterranea*) in the Ivory Coast (Fauquet and Thouvenel, 1987).

However, seed transmission of the virus has not been detected by stringent tests in the following cases: French beans and soybeans in Brazil (Costa et al., 1983), peanuts and soybeans in India (Iizuka et al., 1984), cowpeas and soybeans in Nigeria and soybeans and peanuts in Indonesia (Horn et al., 1991). Seed transmissibility of the virus is thus probably dependent on the interaction between virus strain, plant genotype, duration of infection and, possibly, environmental conditions.

Pest Importance

Although the virus has a wide geographical distribution in Africa, Asia, Oceania and South America, its effect on the growth and yield of infected plants has been rarely studied.

CPMMV was reported to be of minor importance in cowpea crops in Papua New Guinea (Philemon, 1987), in mung beans and French beans (*Phaseolus vulgaris*) in Tanzania (Mink and Keswani, 1987), and in French beans and soybeans in Brazil (Costa et al., 1983). By contrast, the virus can cause yield losses of 64 to 80% in peanuts in Kenya (Bock et al., 1976, 1977). It also causes conspicuous leaf chlorosis and stunting, but unstated yield losses, of infected peanuts, soybeans, bambara groundnuts (*Vigna subterranea*) and winged beans (*Psophocarpus tetragonolobus*) elsewhere, including the Ivory Coast, India and Indonesia (Fauquet et al., 1979; Fortuner et al., 1979; Dubern and Dollet, 1981; Thouvenel et al., 1982; Fauquet and Thouvenel, 1987; Saleh et al., 1989; Reddy, 1991).

Symptoms/Signs

The virus causes conspicuous leaf chlorosis (Fig. 2) and stunting. Viral infection induces brush-like or falcate inclusions within infected plants (Fig. 1) that are probably composed mostly of aggregated virus particles.

Known Hosts

Although most of its natural hosts are leguminous species, CPMMV also occurs naturally in tomatoes in Israel and Nigeria. Isolates of the virus are readily sap-transmissible experimentally to many species of the Fabaceae, and also to some species of the Amaranthaceae, Aizoaceae, Asteraceae, Chenopodiaceae, Cucurbitaceae, Pedaliaceae, Scrophulariaceae, Solanaceae and Sterculiaceae (CABI, 2004).

Figure 2. Mosaic symptoms of CPMMV on soybean. Photo courtesy of Mitsuro Kameya-Iwaki (CABI, 2004).

Major hosts

Arachis hypogaea (peanut), *Glycine max* (soybean), *Lycopersicon esculentum* (tomato), *Phaseolus vulgaris* (common bean), and *Vigna unguiculata* (cowpea)

Minor hosts

Calopogonium mucunoides (calopo), *Mucuna pruriens* (buffalobean), *Phaseolus lunatus* (lima bean), *Phaseolus radiata* (black gram), *Psophocarpus tetragonolobus* (winged bean), *Vicia faba* (broad bean), and *Voandzeia*

subterranea (bambara groundnut)

Wild hosts
Centrosema pubescens (centro), *Desmodium tortuosum* (Florida beggarweed), *Stylosanthes gracile*, and *Tephrosia villosa*

Known Distribution
When it was first described in 1973, this virus was thought to be of only local, and possibly minor, importance in Ghana (Brunt and Kenten, 1973). However, a disease of peanuts, described as 'Ngomeni mottle' (Storey and Ryland, 1957) and since shown to be induced by CPMMV (Bock et al., 1976), occurred in Kenya at least 16 years previously. The virus has possibly also been long present but unrecognized in other countries.

Although no detailed surveys have been made to determine the extent of its geographical distribution, the virus is known to have a wide distribution in countries of Africa (Burkina Faso, Ivory Coast, Egypt, Ghana, Kenya, Malawi, Mozambique, Nigeria, Sudan, Swaziland, Tanzania, Togo, Uganda, Zambia), Asia (India, Indonesia, Israel, Japan, Jordan, Malaysia, Thailand, and Yemen), Oceania (Fiji, Papua New Guinea, the Solomon Islands) and South America (Brazil) (CABI, 2004).

Potential Distribution Within the US
Information is not available at this time.

Survey
There are no specific survey methodologies established for CPMMV. The disease has been traditionally detected based on the examination of the typical symptoms. Because symptoms are not specific for CPMMV, inoculation of indicator plants, ELISA, electron microscopy and/or PCR are necessary to confirm the presence of CPMMV.

Key Diagnostics
The virus is readily transmitted by mechanical inoculation of sap from infected crop plants to diagnostic herbaceous host species, the reactions of which are as follows:

Arachis hypogaea (peanuts) - a few local necrotic lesions, rings or line patterns, and chlorosis, rolling and veinal necrosis of systemically infected leaves. Plants are severely stunted.

Beta vulgaris - a few fawn necrotic local lesions, but no systemic infection.

Cajanus cajan (pigeon peas) - severe chlorosis and distortion of systemically infected leaves and stunting of plants.

Chenopodium quinoa - numerous local chlorotic or necrotic lesions, but no systemic infection.

Glycine max (soybeans) - conspicuous chlorosis of systemically infected leaves and, sometimes, apical chlorosis.

Phaseolus vulgaris (beans) - chlorotic spotting of systemically infected leaves.

The virus is best identified by serological methods of which enzyme-linked immunosorbent assay (ELISA) is the most useful (Antignus and Cohen, 1987; Mali et al., 1989; Mink and Keswani, 1987); this method is also effective when using mixed antisera when screening for several viruses (Hampton et al., 1992). Using ELISA detection methods, CPMMV, however, could not be detected in seeds from 60 cowpea pre-introductions from Botswana, India and Kenya (Gillaspie et al., 1995), in 4144 seeds harvested from seven CPMMV-infected soybean genotypes or in 214 seeds collected from CPMMV-infected peanut plants (cv. Gajah) (Horn et al., 1991).

Immunosorbent electron microscopy is also a very useful diagnostic procedure (Brunt et al., 1983; Gaspar et al., 1985). More recently, a polymerase chain reaction (PCR) procedure has been developed for the rapid and sensitive detection of CPMMV and other carlaviruses (Badge et al., 1996).

Bean Pod Mottle Virus (BPMV)

Scientific Name
Bean Pod Mottle Comovirus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Zaumeyer and Thomas first described Bean pod mottle virus (BPMV) in 1948 on *Phaseolus vulgaris* in Charleston, South Carolina. In 1948, the virus was noted to be readily transmitted mechanically, and the experimental host range included several varieties of all groups of snap and dry beans. In further exploration of the BPMV host range, 25 species including 20 genera of plants were evaluated for susceptibility. In this test, some varieties of lima bean and soybean were determined to be susceptible. BPMV was identified as a soybean problem in the field in 1951 in Arkansas.

BPMV is a member of the genus Comovirus in the family Comoviridae. Like other comoviruses, BPMV has a bipartite positive-strand RNA genome consisting of RNA-1 and RNA-2, which are separately encapsidated in isometric particles 28 nm in diameter. Total genome size 12.8 kb.

Biology and Ecology
BPMV is efficiently transmitted in nature within and between soybean fields by several leaf feeding beetles. Studies have established the bean leaf beetle, *Cerotoma trifurcata*, as the primary vector of BPMV. Other vectors include: *Colaspis brunnea* (grape colaspis), *Colaspis lata, Diabrotica balteata* (banded cucumber beetle), *D. undecimpunctata howardii* (spotted cucumber beetle), *Epicauta vittata* (striped blister beetle), and *Epliachna varivestis* (Mexican bean beetle). More recently, *Diabrotica virgifera virgifera* (western corn root worm) and *Odontota horni* (soybean leaf miner) have been identified as potential vectors (Werner et al., 2002).

Numerous efforts have failed to demonstrate seed transmission of BPMV in soybeans. However, there are two reports of a low level (<0.1%) seed

transmission of BPMV (Lin and Hill, 1983; Ross, 1986). BPMV is stable, easily transmitted mechanically, and present at relatively high levels in seed coats from BPMV-infected plants.

Pest Importance

BPMV is widespread in the major soybean growing areas in the southern and southeastern U.S. A severe outbreak of BPMV in the north central and northern Great Plains states is currently causing serious concerns to soybean growers and to the soybean industry in this region (Giesler et al., 2002). The deleterious effects of BPMV not only reduce yield but also reduce seed quality, as seeds from infected plants may be discolored. Furthermore, BPMV predisposes soybeans to *Phomopsis spp.* seed infection, a major cause of poor seed quality in soybean.

Yield loss to BPMV is generally related to time of infection; early infection can result in severe losses. Virus transmission through seed is very low, generally less than 0.01 %. Like SMV, high temperatures limit BPMV symptom expression, whereas cool temperatures enhance development of leaf symptoms. BPMV is transmitted by several species of leaf feeding beetles, including bean leaf beetle. Overwintering beetles probably acquire the virus from wild legume weed species. No varieties with resistance are available, but varieties do vary with regard to tolerance. Mixed infections of BPMV and SMV can result in severe stunting or death of the plant.

Symptoms/Signs

Soybean response to BPMV infection varies. Plant symptoms range from a mild chlorotic mottling of foliage (Fig. 1) to a severe mosaic. With the most obvious symptoms appearing on young leaves. Depending on the soybean variety, BPMV may cause terminal necrosis and death. BPMV delays maturity of soybean stems, causing ‘green stem’. Green stem syndrome is the condition where the stems of mature plants remain green and leathery making harvest difficult. The pod mottling symptom that is prominent in snap bean is not prominent in many soybean cultivars due to pubescence, but it does appear in some. Soybeans infected with BPMV may produce seed with mottled seed coats (Fig.

Figure 1. Chlorotic mottling on soybean. Photo courtesy of O.W. Bartnett, North Carolina State University.

2). The mottling originates at the hilum and is also referred to as ‘bleeding hilum’, since hilum color appears to bleed from its normal zone. The disease can cause reductions in soybean yield and seed quality. The severity of symptoms is related to the virus strain, soybean variety, and how early the plant is infected.

BPMV interacts synergistically with the potyvirus soybean mosaic virus (SMV), causing drastic reductions in yield and seed quality. It is prudent to use SMV-resistant cultivars in regions where BPMV is endemic.

Figure 2. Mottled seeds. Photo courtesy of Marlin Rice, Iowa State University.

Known Hosts

Cassia occidentalis, *Catharanthus roseus*, *Catharanthus roseus*, *Chenopodium quinoa*, *Desmodium paniculatum*, *Lens culinaris*, *Phaseolus vulgaris*, *Glycine max* (soybean), *Phaseolus lunatus* (lima bean), *Lespedeza* spp., *Mucuna deeringianum*, *Stizolbium deeringianum*, *Trifolium incarnatum,* and *Vigna* spp. have been reported as hosts.

Known Distribution

The virus is known to occur in the U.S. and Canada.

Potential Distribution Within the US

Until recently, BPMV was confined to the southern U.S., including North and South Carolina, Kentucky, Mississippi, Virginia, Lousiana, and Arkansas. The virus has recently been reported in the north central region of the U.S., including Iowa, Illinois, Indiana, Kansas, Nebraska, Ohio, South Dakota, and Wisconsin. BPMV is likely present in all soybean-producing states, but documentation is incomplete (Giesler et al., 2002).

Survey

The virus is currently surveyed for using a visual survey of plant symptoms. The virus must be confirmed using hot inoculations and/or ELISA.

Key Diagnostics

Diagnostic hosts include:

Glycine max: severe mottling and malformation of leaves, pods and seed coats.

Phaseolus vulgaris: cvs Black Valentine, Bountiful, Tendergreen, Cherokee Wax) severe leaf mottling, malformation.

Mucuna deeringianum: mottling.

Trifolium incarnatum: mottling.

Lespedeza striata: mottling.

The recent work of Gu et al. (2002) has revealed at least two genetically distinct BPMV subgroups, I and II. The two subgroups can be clearly distinguished by nucleic acid hybridization analysis.

ELISA testing has been used to positively detect BPMV in soybean plants, seed, and beetles (Krell et al. 2003; Shahraeen and Ghotbi, 2005). ELISA test are available commercially (e.g. Agdia).

Indonesian Soybean dwarf virus (ISDV)

Scientific Name
Indonesian Soybean Dwarf Luteovirus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Little information is currently available on Indonesian soybean dwarf virus (ISDV). It was first reported in *Glycine max* from Bogor, Indonesia by Iwaki *et al.* (1980).

ISDV virions are isometric single stranded, positive sense RNA without an envelope. Virions are 26 nm in diameter, rounded in profile, without a conspicuous capsomere arrangement. Virions are found in the phloem of susceptible plants.

ISDV is transmitted by an aphid vector, *Aphis glycines* in a persistent manner. The virus is retained when the vector molts, but the virus does not multiply in the vector. The virus is not transmitted by mechanical inoculation or contact between plants. It is not transmitted by seed or by pollen (Brunt et al., 1996).

Pest Importance
Information is not available at this time.

Symptoms
On soybean, this virus causes stunting (dwarfing) with shortened petioles and internodes, mottling, and leaf malformation. Leaves are often rolled.

Known Hosts
Major hosts
Glycine max (soybean)

Known Distribution
The virus is known to be present in Indonesia and Thailand (CABI, 2004).

Potential Distribution Within the US

Information is not available at this time.

Survey
Information is not available at this time.

Key Diagnostics
Information is not available at this time.

Mung Bean Yellow Mosaic Virus (MYMV)

Scientific Name
Mung bean yellow mosaic bigeminivirus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Mung bean yellow mosaic virus (MYMV) particles are geminate, paired particles measuring approximately 30 x 18 nm. The particles are stable in 2% sodium phosphotungstate (pH 3.5) or 2% uranyl acetate without fixation. Particles contain two circular single-stranded DNAs (Morinaga et al., 1990), which account for approximately 20% of particle weight (Ikegami et al., 1985). DNA 1 has 2723 nucleotides (22.43% G, 27.02% A, 20.82% C and 29.70% T) and DNA 2 has 2675 nucleotides (20.56% G, 28.85% A, 19.02% C and 31.55% T). The nucleotide sequence of both DNAs has been determined (Moringa et al., 1993). Particles contain one polypeptide of 28.5 kDa, estimated by SDS/PAGE (Ikegami et al., 1995).

The ssDNA from purified virus particles and the circular dsDNA of genome length isolated from infected plants infected with a Thai isolate are both infective by mechanical inoculation. Cloned linear dsDNA is also infective and DNA segments 1 and 2 are both required for infectivity to mung bean plants (Morinaga et al., 1990). DNA 1 can replicate independently in inoculated non-host tobacco protoplasts (Cheng and Ikegami, 1991).

DNA 1 and DNA 2 have little sequence similarity, except for a region of approximately 200 bases, which is almost identical in the two molecules (Morinaga et al., 1993). Within this common region, there is a 34-base sequence capable of forming a stable hairpin structure (Morinaga et al., 1993). Analysis of open reading frames revealed nine potential coding regions for proteins with a MW of >10.0 kDa, six in DNA 1 and three in DNA 2 (Morinaga et al., 1993). The coat protein gene (1R1) lies on DNA 1 and is encoded by the strand which occurs in the virus particles (Ikegami et al., 1995).

Biology and Ecology

MYMV is transmissible by the whitefly, *Bemisia tabaci* (Nair and Nene, 1973). Acquisition and inoculation by adults can be effected in a minimum of 15 minutes (Nair and Nene, 1973). A single viruliferous adult can transmit the virus. The virus persists in male and female adults for a maximum of 3 and 10 days, respectively, but not throughout the life cycle of the vector (Ranthi and Nene, 1974).

The nymphal stage can acquire the virus (Ranthi and Nene, 1974). Adults acquire the virus from inoculated *Vigna mungo* plants 1 to 3 days before the appearance of symptoms (Ranthi and Nene, 1974). The virus is not transmitted through the eggs of *B. tabaci* (Ranthi and Nene, 1974). The virus is not seed transmitted in mung bean or soybean (Nair and Nene, 1973).

Pest Importance

MYMV causes serious yield losses in pulse crops in India. The virus caused 75.8, 51.8 and 15.2% reduction in yield of the soybean cv. Bragg when infected at the pre-bloom, bloom, and post-bloom stages of growth (Dhingra and Chenulu, 1985). There was a 62.9% maximum growth reduction and 83.9% maximum yield loss for mung bean crops in which symptoms appeared 20 days after sowing. Inoculation of urd bean plants of up to 3 weeks old results in the complete loss of seed yield.

Symptoms/Signs

Mung bean: Small yellow specks along the veinlets of leaves, which spread over the lamina to produce yellow mosaic symptoms. The pods become thin and curl upwards.

Black gram: Two types of yellow mosaic symptom are induced depending on the variety; 'yellow mottle', a generalized yellowing of the leaves and 'necrotic mottle', in which yellowing is restricted to small spots which become necrotic.

Figure 1. Yellow mosaic symptoms on mung bean leaves at late stage of infection. Photo courtesy of M. Ikegami (CABI, 2004)

In India, the virus causes more severe yellow mosaic disease in black gram than in mung bean. However, in Thailand, the disease is common in mung bean but is seldom observed in black gram under natural conditions.

Diagnostic Species:

Soybean: Small, yellow specks initially develop along the veinlets and later coalesce to produce yellow mosaic.

Mung bean: Irregular chlorotic spots along the veinlets, which develop into yellow mosaic. The first emerging trifoliate leaves often show severe downward curling.

French bean (*Phaseolus vulgaris*): Young, systematically infected leaflets show downward curling without yellow mosaic. Occasionally, irregular chlorotic spots develop.

No local lesion host is known.

Known Hosts

Mung bean and black gram were long thought to be the only natural hosts of this virus. However, more recently it has been reported in India infecting pigeonpea (*Cajanus cajan*) (Mandal et al., 1998) and soybean (*Glycine max)* (Dantre et al., 1996; Bhagabati and Mahato, 1999).

Major hosts

Vigna mungo (black gram) and *Vigna radiata* (mung bean)

Minor hosts

Cajanus cajan (pigeon pea) and *Glycine max* (soybean)

Experimental Hosts

MYMV from Thailand has been transmitted by mechanical inoculation to *Canavalia ensiformis*, soybean (*Glycine max)*, *Phaseolus angularis*, *P. vulgaris*, *Vigna mungo* and *V. radiata* (Fabaceae) (Honda et al., 1983). The virus first reported from India has not been transmitted mechanically but has been transmitted by the whitefly vector, *Bemisia tabaci*, to several species in Fabaceae (Nariani, 1960) and also to *Brachiaria ramosa* (Poaceae) and C*osmos bipinnatus*, *Eclipta alba* and *Xanthium strumarium* (Asteraceae) (Nene, 1973; Rathi and Nene, 1974). Another isolate of MYMV from pigeonpea in India has a slightly more extensive host range (Mandel et al., 1997).

Known Distribution

The virus is only known to occur in Asia (Bangladesh, India, Pakistan, Philippines, Sri Lanka, and Thailand) and Papua New Guinea.

Potential Distribution Within the US

Information is not available at this time.

Survey

There are no specific survey methodologies established for MYMV. The disease has been traditionally detected based on the examination of the typical

symptoms on leaves. Because symptoms are not specific for MYMV, ELISA or DNA probes are necessary to confirm the presence of MYMV.

Key Diagnostics

The experimental host range of MYMV (Indian isolate) is narrow. *Cajanus cajan*, *Glycine max*, *Macroptilium lathyroides*, *Macrotyloma uniflorum, Phaseolus vulgaris* cv. Manitov, *P. aconitifolia*, *Vigna radiata*, *P. aureus*, *V. unguiculata* and *V. mungo* are susceptible to infection, but *Hibiscus esculentus* and *P. lunatus* are not (Nariani, 1960; Nene, 1972).

An antiserum to the Thai isolate of MYMV, with a titer of 1/512 in gel double diffusion tests, has been used to detect MYMV (Ikegami and Shimizu, 1988). ELISA has been used more recently for virus identification (Mandal et al., 1997).

Cloned probes have been prepared to each of the two genome DNAs; those for either DNA can be used in spot hybridization tests to detect infection by MYMV (Morinaga et al., 1990). Probes for DNA B have been used in India to detect MYMV in plants and whiteflies (Rani et al., 1996; Mandal et al., 1997).

Peanut Stunt Virus (PSV)

Scientific Name
Peanut Stunt Cucumovirus

Common Name(s)
Peanut stunt virus, black locust true mosaic virus, clover blotch virus, groundnut stunt virus, peanut common mosaic virus, robinia mosaic virus

Type of Pest
Plant pathogenic virus

Reason for inclusion in manual

Pest Description
Peanut stunt virus (PSV) is a member of the genus Cucumovirus in the family Bromoviridae. Other members of the genus are tomato aspermy virus (TAV) and the type member cucumber mosaic virus (CMV). The chemical composition of PSV is 16% nucleic acid and 84% coat protein. Like other cucumoviruses, PSV has a tripartite genome of positive-strand RNAs, designated RNAs 1, 2, and 3. The RNAs are packaged in isometric particles of about 28 nm in diameter (Fig. 1). RNAs 1 and 2 encode the 1a and 2a proteins, respectively, which along with host components are required for replication. RNA 3 is dicistronic and encodes a putative movement protein (MP) and the coat protein (CP), which is expressed from subgenomic RNA 4. RNA 4 is packaged together with RNA 3. A small overlapping gene (2b), encoded by RNA 2, is present in all cucumoviruses sequenced to date and is expressed through subgenomic RNA 4A. CMV 2b

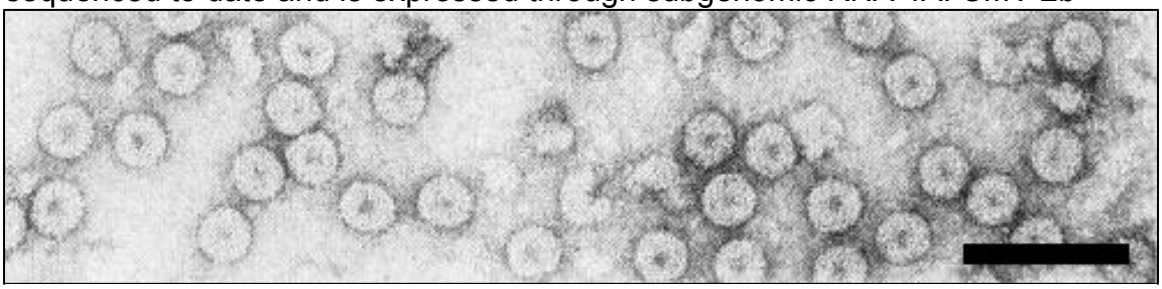


Figure 1. Peanut stunt virus particles from a purified preparation stained with 1% uranyl acetate. Bar represents 100 nm. Photo courtesy of S. A. Tolin.

protein, essential for systemic virus spread in cucumber, has been identified as a suppressor of posttranscriptional gene silencing, and thus a pathogenicity determinant.

Strains of PSV have been classified into two major subgroups (subgroups I and II) based on serology and percent nucleotide sequence identity. Furthermore, a naturally occurring reassortment (PSV-BV-15) between the two subgroups has been reported and characterized at the molecular level (Hu and Ghabrial, 1998).

Biology and Ecology

PSV is transmitted in nature by insect vectors belonging to the Aphididae, such as *Aphis craccivora* (cowpea aphid), *A. spiraecola (*spiraea or apple aphid), and *Myzus persicae* (green peach aphid), but not by *A. gossypii* (melon or cotton aphid). A study showed that one field isolate of PSV was not transmitted by *A. glycines* (soybean aphid) (Clark and Perry, 2002). It is transmitted in the non-persistent manner. PSV can also be transmitted by mechanical inoculation. PSV is transmitted in a small percentage of peanut seeds (about 0.1%) and soybeans (3 to 18%). White clover is an important overwintering source of the virus for infection of peanuts, tobacco, and beans in the U.S. Infected peanut seeds, however, are not though to play a role in the spread of disease since only seeds too small for planting are infected at a high enough rate to act as a source of inoculum. The black locust mosaic tree (*Robinia pseudoacacia*) was found to be a primary source of PSV in China.

Seeds from plants that were infected early appear misshapen, frequently with a split pericarp wall, and have poor viability. As seedlings from such seeds emerge late and grow poorly, seed transmission is not considered an important factor in the spread of disease. Seed transmission is also not considered of importance when perennial hosts are present. Seed transmission of PSV in hosts other than peanuts and soybeans has not been reported.

Pest Importance

PSV is an economically important pathogen of legumes worldwide. PSV may cause disease in peanut, cowpea, tobacco, clover, soybean, and snapbean. Following early infection in peanut, plants are stunted and may never grow beyond a few inches in height and width.

In the 1960s, PSV was a problem in peanut in Virginia, North Carolina, and Georgia, but it is not of economic importance for peanut production in the U.S. However, PSV causes a high incidence of peanut stunt disease in Hebei, Henan, and Lianing provinces in China. In greenhouse studies, PSV has been implicated as a weakening factor in white clover plants, because it renders them more susceptible to injury and death from environmental stress and diseases. Alternately, plants may die as a direct result of PSV infection.

The virus can overwinter in wild or forage legumes (e.g. clovers, alfalfa, and lespedeza) and then spread to other crops in the spring by aphids that carry the virus in their mouthparts after feeding on infected plants. Peanut yield and value are reduced by peanut stunt virus because of a decrease in numbers of fancy pods, extra large kernels, and sound mature kernels.

Symptoms/Signs

Symptoms of PSV vary depending on the host plant and strain of the virus. Symptoms shown by naturally infected plants persist. In peanuts (*Arachis hypogaea*), there are various degrees of stunting, shortening of petioles, reduced leaf size, mild mottling, and malformation of pods (Fig. 2). Leaves from plants infected with peanut stunt virus are malformed and curl up at the edges. Infected leaves may be paler green and/or yellowed. The fruit of plants infected with the virus are commonly split open to expose seed. In beans (*Phaseolus* spp.), chlorotic mottling and mosaic occurs on trifoliate leaves; in some cultivars trifoliate leaves are elongated and misshapen. In clover, (*Trifolium* spp.), a faint to severe mosaic or interveinal chlorosis (Fig. 3) occurs sometimes with necrosis and leaf malformation. Nodulation ratings are generally lower (fewer nodules, smaller nodules, abnormal color and shape) in white clover plants infected with

Figure 2. Symptoms of peanut stunt virus on peanut (bottom). Top of photo shows a healthy peanut plant. Photo courtesy of North Carolina State University, Department of Plant Pathology.

PSV than in non-infected plants. In tobacco (*Nicotiana spp.*), there is a severe mottling, general chlorosis, chlorotic spots, and oak-leaf line patterns. In lupine (*Lupins luteus* and *L. albus*), deformation and distortion of the leaves and flowers occurs and plant dwarfing may occur.

Known Hosts

Figure 3. Peanut stunt virus symptoms on arrowleaf clover. Photo courtesy of O.W. Barnett, North Carolina State University.

Apium gravelens (celery), *Arachis hypogaea* (peanut), *Coronilla varia* (crown vetch), *Datura stramonium* (jimsonweed), *Glycine max* (soybean), *Lupinus albus* (white lupine), *Lupinus luteus* (yellow lupine), *Lycopersicon esculentum* (tomato), *Medicago sativia* (alfalfa), *Nicotiana tabacum* (tobacco), *Phaseolus* spp. (beans), *Pisum sativum* (peas), *Tephrosia* (hoary pea), *Trifolium* spp. (clover), *Vicia faba* (broad bean), *Vigna angularis* (adzuki bean), *Vigna unguiculata* (cowpea).

PSV naturally infects several leguminous hosts (beans, soybeans, cowpea, crown vetch, tephrosia, and lupines) and one or more species of Solanaceae (tobacco, tomato, and jimsonweed). Its experimental host range includes many dicotyledonous species in the Fabaceae, Chenopodiaceae, Compositae, Cucurbitaceae, and Solanaceae.

Known Distribution

The virus is known to be present in **Asia:** China, Republic of Georgia, Japan, Korea, **Europe:** France, Hungary, Italy, Poland, Spain, Morocco, **Africa:** Sudan, and **North America:** the U.S.

Potential Distribution Within the US

This relatively new virus was first found in peanut in Virginia and North Carolina in 1964, when it caused severe loss in crop yield and value. PSV is also present in Alabama, Arkansas, Florida, Illinois, Iowa, Kentucky, and Washington.

Survey

There are no specific survey methodologies established for peanut stunt virus. The disease has been traditionally detected based on the examination of the typical symptoms on leaves coupled with the stunting symptom. Because symptoms are not specific for PSV, ELISA or PCR are necessary to confirm the presence of PSV.

In one study surveying for five viruses, including peanut stunt, soybean plants showing virus-like symptoms were dug between July and September. A total of 18 composite samples of 10 leaflets from asymptomatic soybean plants were also collected, with each leaflet from a different plant. Plants were placed in plastic bags, watered, transported on ice, and stored at 4 °C prior to virus testing and transmission studies, including mechanical and aphid transmission and serological testing (Clark and Perry, 2002).

Key Diagnostics

Diagnostic indicator plants are commonly used in virus identification; however, they are not always reliable because different strains of the same virus may cause different reactions on the same plant species or have different host ranges, or different viruses may cause similar reactions on the same plant. The experimental host range of PSV is wide. The following plants are usually used as diagnostic hosts for PSV:

Vigna unguiculata (cowpeas): chlorotic local lesions, systemic vein clearing and severe epinasty.

Chenopodium amaranticolor and *C. quinoa:* chlorotic local lesions and systemic spotting.

Phaseolus vulgaris: chlorotic or necrotic local lesions and systemic chlorotic mottling or mosaic: the severity of the symptom depends on the cultivar (e.g. in ‘Bountiful’ the systemic symptoms are elongated, misshapen trifoliate leaves).

Pisum sativum: systemic chlorotic mottling and stunting.

Nicotiana tabacum: light green and yellow rings 5 to 10 mm in diameter on inoculated leaves and chlorotic areas on young systemically infected leaves.

Serology is perhaps the most easily and widely accepted method of identifying viruses. Several serological techniques are useful for identifying PSV, including immunodiffusion tests and enzyme-linked immunosorbent assay (ELISA). However, ELISA is more sensitive and hence more widely used for the detection of PSV in plants and seeds. Both double antibody sandwich (DAS)-ELISA and an indirect ELISA procedure can be used to detect PSV (Anderson et al., 1991; McLaughlin et al., 1984). Optimum concentrations/dilutions of the antisera have to be determined experimentally in order to avoid non-specific background

reactions. Tests in which the optical density values (A 405 nm) are more than double the negative controls are scored as positive.

More recently PSV can be differentiated from four other legume infecting viruses (alfalfa mosaic, bean yellow mosaic, clover yellow vein, and cucumber mosaic viruses) in one single multiplex polymerase chain reaction test (Bariana et al., 1994). Peanut stunt can be detected and differentiated from peanut stripe, peanut mottle, and cucumber mosaic virus using reverse transcription polymerase chain reaction assays (Dietzgen et al., 2001).

Red Clover Mottle Virus (RCMV)

Scientific Name
Red Clover Mottle Comovirus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Red clover mottle virus (RCMV) was first reported in *Trifolium pretense* from the United Kingdom (Sinha, 1960).

RCMV is a member of the comovirus group of plant viruses. Its genome consists of two molecules of positive-strand RNA, RNA 1 and RNA 2, which are separately encapsidated in isometric particles about 28 nm in diameter (Fig. 1). The virions are not enveloped, angular in profile; without a conspicuous capsomere arrangement.The virus occurs in Europe and has been independently isolated in several countries including England, Germany, The Netherlands, Slovakia, Sweden, and Ukraine (Lapchick et al., 1998).

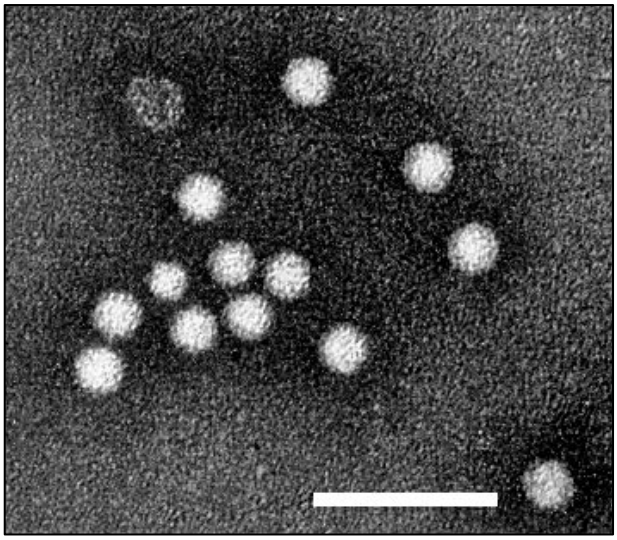


Figure 1. Virus particles from a purified preparation, stained with phosphotungstate. Bar represents 100 nm. Photo courtesy of Rothamsted Experimental Station.

Total genome length is 9570 nucleotides. Virions contain 36% nucleic acid (B), or 25% nucleic acid (M), or 0% nucleic acid (T). Virions contain two segments of linear single stranded RNA. Three structural virion proteins found. Virions contain 64% protein (B), or 75% protein (M), or 100% protein (T). Protein size of the

largest 40000 Da. Protein size of 2nd largest 22200 Da. Protein size of 3rd largest 18300 Da.

In broad bean or French bean sap, the thermal inactivation point (10 min) is between 70 and 75°C and infectivity is retained at 20°C for more than 2 weeks. In pea sap, the dilution endpoint may be up to 10^{-6}.

Biology and Ecology

RCMV is transmitted by the weevil vectors, *Apion aricans* and *A. varipes*. The virus is also transmitted by mechanical inoculation.

Pest Importance

Information is not available on yield or crop loss due to RCMV.

Symptoms/Signs

Apparently all tissues are infected, including roots. Inclusion bodies are not reported, and the distribution of virus within the cell is not known. RCMV is reported to cause mosaic, mottling, and stunting in red clover (*Trifolium pretense*) (Fig. 2).

Known Hosts

Chenopodium amaranticolor (lambsquarters), *Chenopodium quinoa* (quinoa), *Glycine max* (soybean), *Gomphrena globosa*, *Lathyrus odoratus* (sweet-pea), *Medicago sativa* (alfalfa), *Melilotus albus* (yellow sweetclover), *Nicotiana tabacum* (tobacco), *Phaseolus vulgaris* (bean), *Pisum sativum* (pea), *Trifolium hybridum* (alsike clover), *Trifolium incarnatum* (crimson clover), *Trifolium pratense* (red, purple clover), *Trifolium repens* (white clover), *Trifolium subterraneum* (sub clover), *Vicia faba* (broad bean), *Vicia sativa* (vetch), and *Vigna unguiculata* (cowpea) (Brunt et al., 1996).

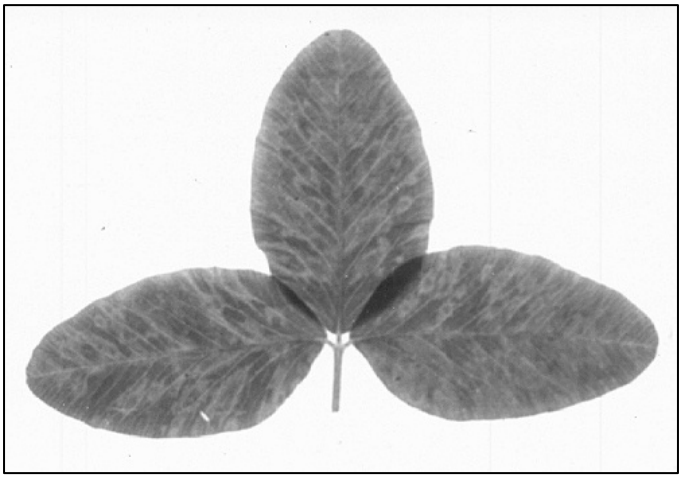


Figure 2. Systemically infected leaf of red clover showing mottling, chlorotic rings and spots. Photo courtesy of Rothamsted Experimental Station.

Known Distribution

RCMV has been reported from most parts of Europe, except the south (CABI, 2004).

Potential Distribution Within the US
Information is not available at this time.

Survey
There are no specific survey methodologies established for red clover mottle virus. The disease has been traditionally detected based on the examination of the typical symptoms on leaves coupled with the stunting symptom. Because symptoms are not specific for PSV, gel double diffusion precipitin tests, or inoculation of diagnostic hosts are necessary for diagnosis.

Key Diagnostics
Diagnostic hosts include:

Gomphrena globosa and *Vicia faba*: necrotic local lesions.

Chenopodium amaranticolor and *C. quinoa*: chlorotic local lesions becoming necrotic.

Glycine max: systemic mosaic, dwarfing.

Lathyrus odoratus: latent infection.

Phaseolus vulgaris: necrotic local lesions (14/82 cultivars).

Pisum sativum: chlorosis, stunting.

Trifolium pretense: mosaic, dwarfing.

Vicia sativa: necrotic and chlorotic local lesions, leaf abscission (Brunt et al., 1996).

Assay (local lesion) hosts include: *Gomphrena globosa*, *Chenopodium amaranticolor*, *C. quinoa*, *Phaseolus vulgaris*, and *Trifolium incarnatum.*

The virus is a good immunogen. In gel double diffusion precipitin tests, agarose (up to 1%) gives better results than agar. Several bands of precipitate may form in these tests with either rabbit hyperimmune sera or mouse immune ascitic fluid; the reason for this is not known.

Red clover mottle virus may be confused with alfalfa mosaic virus in some hosts in which it causes similar symptoms. However, unlike red clover mottle virus, alfalfa mosaic virus is transmitted by aphids, infects *Nicotiana* species and has distinctive bacilliform particles.

Soybean Dwarf virus (SbDV)

Scientific Name
Soybean dwarf luteovirus

Common Name(s)
Soybean dwarf virus - leaf yellowing strain (SDV-Y), *subterranean clover red leaf luteovirus, subterranean clover red leaf virus, soybean dwarf virus - dwarfing strain* (SDV-D)

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Soybean dwarf virus (SbDV) was first described by Tamada et al. (1969). SbDV exists as a group of closely related strains. Two strains, referred to as the dwarfing strain (SDV-D) and the yellowing strain (SDV-Y) (Fig. 1), have been identified by Tamada (1973). Subterranean clover red leaf virus was reported as a separate virus from Australia and New Zealand, but is considered to be a strain of SbDV (here it is called SDV-SCRLV) based on the similarities of the vector species, host range, symptomatology and serological reactions (Ashby and Kyriakou, 1982; Johnstone and McLean, 1987).

Figure 1. Interveinal chlorosis (yellowing) caused by the SDV-Y strain. Photo courtesy of Tetsuo Tamata (CABI, 2004).

SbDV is a member of the family Luteoviridae. The name luteovirus is derived from the Latin

'luteus' (yellow) and the symptoms caused by these viruses are, in general, yellowing. Other characteristic symptoms are reddening, rolling, curling and brittleness of the leaves. Luteoviruses are phloem-restricted; they are transmitted by aphids in a persistent (circulative, non-propagative) manner but are not transmitted either by mechanical inoculation or through seeds. The member viruses of the luteovirus genus show evolutionary relationships to members of the genera sobemovirus and carmovirus. Based on the genome structure and organization, luteoviruses are now divided into two subgroups I and II. SbDV belongs to subgroup I (Mayo and Ziegler-Graff, 1996).

The virus particles are 25 nm in diameter, hexagonal in outline and have no envelope or surface features (Tamada and Kojima, 1977; Ashby and Kyriakou, 1982; Hewings et al., 1986). Particles contain a single molecule of infectious, linear, positive-sense ssRNA. The genome size is 5861 nucleotides for the Tas isolate of SDV-SCRLV (Rathjen et al., 1994).

Biology and Ecology

SbDV is not transmitted through seed (Tamada et al., 1969). SbDV is most efficiently transmitted by the aphid *Aulacorthum solani* in the persistent (circulative) manner. *A. circumflexum* is also an efficient vector of the virus, but is unlikely to be important in nature (Helms et al., 1983). Furthermore, one or more strains of SbDV were reported to be efficiently transmitted by *Acyrthosiphon pisum* in Australia, the U.S., Japan, and Syria (Makkouk et al., 1997). Such *Acyrthosiphon pisum*-specific strains are less or not efficiently transmitted by *A. solani*. Thus, there are vector-specific strains of SbDV.

SbDV was found not to be transmitted by other aphid species tested, *Aphis glycines*, *Aphis craccivora*, *Aphis gossypii*, *Myzus persicae*, *Acyrthosiphon kondoi*, *Macrosiphum euphorbiae* and *Therioaphis trifolii* f. sp. *maculata* (CABI, 2004).

In general, the virus is acquired by phloem feeding of aphids, enters the haemocoel of the aphid via the hind gut, circulates in the haemolymph, and probably enters the accessory salivary gland. Inoculation probably results from transport of virus into the salivary duct and introduction of saliva into the plant during feeding.

Pest Importance

SbDV causes a serious loss of soybeans in Japan. Yield losses in soybeans vary from mild to almost total loss depending on cultivar, virus strain, aphid density, plant age when infected, and environmental conditions. Inoculation tests showed that SDV-Y causes much more severe losses in major soybean cultivars and lines than SDV-D (Tamada, 1975). In naturally infected fields, for example, a 50% level of field infection results in a 40% reduction in yield, mostly through a reduction in the number of pods per plant (Tamada, 1975).

It is unknown whether SbDV is important for soybean production in other countries. Damsteegt et al. (1990) reported that SbDV does not appear to pose an economic threat to soybean production in the U.S.

In Japan, SDV-Y causes a serious damage in common beans (Tamada, 1975). In Australia and New Zealand, SDV-SCRLV causes severe or mild yield loss in legume pastures such as subterranean clover and in grain legumes, such as broad beans, lupines and peas (Wilson and Close, 1973; Johnstone, 1978; Ashby et al., 1979).

Symptoms/Signs

Soybean: SDV-D causes dwarfing. Dwarfed plants show shortened petioles and internodes. Younger leaflets are faintly yellow. Older leaflets are dark green, smaller than normal, thick, brittle, and curl downward. SDV-Y causes slight cupping of very young leaves. Older leaflet margins appear undulate, instead of smooth. Leaflets become rugose or wrinkled, remain smaller than normal (Fig. 2). Older leaflets become thickened and brittle; interveinal yellowing or marginal reddening appears on older leaves. Stunting by SDV-Y is moderate compared to that by SDV-D. Plants affected by SDV-Y have a more open habit than healthy plants. Plants infected by both virus strains show symptoms more severe than those infected with either strain alone, including very rugose leaves. Younger soybeans are more susceptible than older soybeans. Unifoliolate and first-trifoliolate leaf stages are more susceptible than second-trifoliolate leaf stages and older. Younger leaves are also more susceptible than older leaves.

Figure 2. Leaf rugosity and yellowing of naturally infected soybean with SbDV. Photo courtesy of Tetsuo Tamata (CABI, 2004).

Subterranean clover: Diseased plants exhibit leaves that turn bright red as they mature, with smaller leaves than those on healthy plants. Reddening begins at leaflet margins and gradually spreads interveinally towards the midrib. Later, leaf margins become necrotic. Plants are slightly stunted but continue to grow for a year.

Beans: Plants are stunted. Leaves are yellowed, thickened, curl downward, and drop prematurely. Few pods are set.

Broad bean: Symptoms are more obvious in the cooler than the warmer part of the season. Leaves roll upwards about the midrib, interveinal chlorosis is a bright to dull yellow, and texture is thickened and rough. Few pods are set.

Peas: Diseased plants are stunted with top yellowing. Plants become rigid and brittle with shoots proliferating from nodal buds at the plant base. Plants often succumb to secondary fungal root rot. Older leaves exhibit interveinal chlorosis; leaves later turn bright yellow, sometimes with orange tints and veinal necrosis.

The virus particles are restricted to the phloem tissues (i.e. sieve elements, companion and phloem parenchyma cells). Phloem necrosis is seen in some cells of such tissues and causes external symptoms by inhibiting translocation and slowing plant growth.

Known Hosts

More than 50 leguminous and a few non-leguminous species are susceptible to SbDV (Tamada, 1973; Ashby et al., 1979). SbDV strains differ in host range and symptomatology. In general, SDV-Y causes more severe symptoms than SDV-D. The host range of SDV-SCRLV is more similar to that of SDV-Y than to that of SDV-D (Damsteegt et al., 1990). *Astragalus sinicus, Glycine max, Pisum sativum, Crotalaria juncea, C. zanzibarica*, *Lens culinaris*, *Trifolium subterraneum*, *T. dubium*, *T. hybridum*, *T. incarnatum*, *Vicia faba* and *V. sativa* are susceptible to all SbDV strains. *Lupinus* spp., *Phaseolus vulgaris* and *T. repens* are hosts of SDV-Y and SDV-SCRLV, but are immune to infection with SDV-D. *T. campestre*, *T. pratense*, *T. tembense*, *T. tomentosum*, *T. variegatum* and *T. wormskioldii* are susceptible to SDV-D, but not to SDV-Y. Apart from the Fabaceae, *Beta vulgaris*, *Spinacia oleracea* and *Phlox drummondii* are hosts of all virus strains (Damsteegt et al., 1990).

Major hosts

Beta vulgaris var. *saccharifera* (sugarbeet), *Glycine max* (soybean), *Gomphrena globosa* (Globe amaranth), *Lens culinaris* (lentil), *Lupinus* (lupines), *Lupinus albus* (white lupine), *Lupinus angustifolius* (lupine), *Lupinus luteus* (yellow lupine), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Pisum sativum* var. *arvense* (Austrian winter pea), *Trifolium dubium* (yellow suckling clover), *Trifolium fragiferum* (strawberry clover), *Trifolium hybridum* (Alsike clover), *Trifolium incarnatum* (crimson clover), *Trifolium pratense* (purple clover),

Trifolium repens (white clover), *Trifolium subterraneum* (subterranean clover), *Vicia articulata* (oneflowered vetch), *Vicia faba* (broad bean), *Vicia faba* var. *major* (broad bean), *Vicia sativa* (common vetch)

Known Distribution

SbDV was first recognized as a distinct virus of soybeans in the southern areas of Hokkaido, Japan in 1952 (Tamada et al., 1969). Since then the disease has gradually spread in Japan.

SDV-SCRLV was first recorded affecting subterranean clover in Victoria, Australia in 1965 (Kellock, 1971). The virus has been found in the Canterbury region of New Zealand and in all southern Australian states (Helms et al., 1993).

The U.S. strain of SbDV (SDV-US), which was detected in white clover, is widespread in the southeastern U.S. (Damsteegt et al., 1995).

Although SDV-D, SDV-Y and SDV-SCRLV are efficiently transmitted by *Aulacorthum solani*, one or more strains of SbDV which are transmitted specifically by *Acyrthosiphon pisum* are reported to occur in Australia, the U.S. and Japan (CABI, 2004).

Potential Distribution Within the US

The virus is currently present in Alabama, California, Kentucky, Maryland, New York, North Carolina, Pennsylvania, South Carolina, and Virginia.

Survey

To survey for SbDV in the field, visible disease symptoms are used, which includes dwarfing (stunting), leaf rugosing and yellowing. Check soybean fields for plants shorter than normal. Various leaflets may be smaller, curled downward, wrinkled; thick and brittle with younger leaflets yellow, and older leaflets dark green. Older leaflets may show interveinal yellowing. In subterranean clover, look in late winter or early spring for the first symptoms. Leaves redden from the margins inwards. Plants may collapse and rot before the end of the growing season (USDA, 1987).

Key Diagnostics

SbDV can be detected by ELISA (D'Arcy and Hewings, 1986) and by aphid transmission to indicator plants (Tamada and Kojima, 1977). Nucleic acid hybridization (Martin and D'Arcy, 1990) and RT-PCR (Bariana et al., 1994) assays have been developed for the detection of SbDV. ELISA is the most useful method for detection and diagnosis of SbDV. However, not all strains of SbDV are distinguished by ELISA. SDV-D is distinguished by symptomatology and host range from SDV-Y and SDV-SCRLV, which are indistinguishable. In addition, these strains have slightly different dsRNA profiles and molecular weight of coat proteins (Hewings et al., 1986; Smith et al., 1991).

Aphid inoculation tests are also important for diagnosis. For diagnosis of vector-specific strains, the aphid species usually used are *Acyrthosiphon solani*, (*Aulacorthum solani)* (the foxglove aphid), and *Acyrthosiphon pisum* (the pea aphid). Indicator plant species include *Glycine max*, *Lens culinaris*, *Pisum sativum*, *Trifolium incarnatum*, T. subterraneum and *Vicia faba*, which are susceptible to all strains of SbDV, and *Lupinus albus*, *Phaseolus vulgaris*, *T. pratense* and *T. repens,* which are useful for distinguishing SDV-D from SDV-Y (SDV-SCRLV). Symptoms are stunting, chlorosis and interveinal yellowing or marginal reddening of older leaves.

Dwarfing disease of soybeans with symptoms similar to SbDV have been reported from Indonesia (Iwaki et al., 1980) and Nigeria (Rossel and Thottappilly, 1982). The Indonesian virus is transmitted by *Aphis glycines* and is serologically unrelated to SbDV (Iwaki et al., 1980). The vector of the Nigerian soybean dwarfing pathogen has been identified as *Bemisia tabaci*, although the pathogen is serologically related to SbDV (Anon., 1982).

Similar dwarfing diseases of soybeans are also caused by some other persistent (circulative) aphid-borne viruses of legumes including bean leaf roll virus (Ashby, 1984), beet western yellows virus (Duffus and Milbrath, 1977), and milk vetch dwarf virus (Inouye et al., 1968). However, these viruses are distinguished from SbDV by vector species, host range and serological reactions. Bean leaf roll and beet western yellows viruses belong to the luteovirus genus, and milk vetch dwarf virus seems to be an unassigned virus.

Soybean Mosaic Virus (SMV)

Scientific Name
Soybean mosaic potyvirus

Common Name(s)
Soja virus 1, soybean virus 1

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual

Pest Description
Soybean mosaic virus (SMV) particles are usually flexuous filaments, about 750 nm long and 15 to 18 nm wide (Bos, 1972; Demski and Kuhn, 1989). They have helical symmetry with a pitch of 3.4 nm and contain 5.3% single-stranded nucleic acid (RNA) with a MW of 3,250,000 (Hill and Benner, 1980b). The protein coat contains about 2166 subunits of one type of protein with a MW of approximately 28,300 (Hill and Benner, 1980a) or approximately 32,150 or 33,075 according to virus strain. The genome of two strains of the virus is 9588 nucleotides long, and a large open reading frame encodes for a large precursor polyprotein that is proteolytically cleft into nine mature proteins including the viral coat protein (Jayaram et al., 1992). The protein coat contains about 2166 subunits of one type of protein with a MW of approximately 28,300 (Hill and Benner, 1980a).

The virus remains infective in expressed plant sap for 2 to 5 days. It usually has a dilution end point of 0.001 and a thermal inactivation point of 55 to 70°C.

Strain distinction is essential for breeding and selection for resistance. Many isolates of SMV from the seeds of soybean plants in the U.S., introduced from around the world, have been classified into seven strain groups (G1 to G7) based on their differential interactions with five soybean cultivars.

Biology and Ecology
SMV is transmitted non-specifically by aphids in the non-persistent stylet-borne manner. Soybean has not been reported to be colonized by aphids to any extent. In central Illinois, U.S., more than 60 aphid species are transient in soybean

fields each year (Halbert et al., 1981) and large numbers of aphids are reported to alight and briefly probe soybean plants. Spread of SMV is by several aphid species (mostly transient alatae) (Irwin and Goodman, 1981), but only a few species are important vectors of the virus. In India, *Myzus persicae* gave the highest percentage of transmission of four aphid species. Transmissibility of isolates of the pathogen has been shown to differ between vector species (Lucas and Hill, 1980; O'Connell-Ziegler et al., 1986). *M. persicae* optimally transmitted SMV after acquisition probes of 30 to 60 seconds; transmission was lower after acquisition probes of 15 seconds and access times of 15 minutes or more (Irwin and Goodman, 1981). Plant pubescence hinders aphid probing on leaves, limits virus transmission, and delays epidemic build-up in the field (Ren et al., 2000). SMV may also affect the population density and behavior of vector aphids. *Rhopalosiphum maidis* was found to leave SMV-infected soybean plants sooner after probing than healthy plants, which increases the probability of successfully transmitting the virus to other plants (Fereres et al., 1999).

The transfer of non-persistently transmitted viruses, including SMV by aphids, usually occurs over short distances (up to a few hundred meters, particularly downwind and by migrating alatae (Irwin and Goodman, 1981). In the case of aphid starvation after virus uptake, stylet-borne viruses may persist for some hours, and they may be carried over long distances. Long-distance transfer of non-persistent viruses (12 km) by aphids has been recorded for other non-persistently transmitted legume viruses. For SMV, such influx from even farther away cannot be excluded.

SMV was one of the first viruses detected to be transmitted to plants from seed harvested from infected plants (Kendrick and Gardner, 1924). Seed transmission was demonstrated in a field environment by plants developing the disease when grown from infected seeds that were protected from insect-borne inoculum by insect-proof cages. The lack of an overwintering host in soybean production areas of the northern U.S. further suggested the seed to be the primary inoculum source for the pathogen (Hill et al., 1980). Tu (1989) demonstrated transmission rates as high as 75.6%.

Infectious SMV was recovered from embryos and cotyledons but not the testae of mature seeds. It was, however, found in the testae of immature seeds, and non-infectious antigen was detected by serology in testae of mature seeds (Bowers and Goodman, 1979; Iwai et al., 1985). The pathogen was found extensively in the flowers and pods of inoculated plants (Bowers and Goodman, 1979) and was transmitted to seeds through both pollen and ovules (Iizuka, 1973).

Humans play a dominant and quantitative role in the ecology of SMV by deciding to grow soybean, deciding which cultivar to grow, and by choosing the time, extent and way of cultivation. Man also continues to introduce and spread the

virus in commercial seed and in germplasm used for breeding and in breeders' material used for multilocational testing.

Pest Importance

Reports on the impact of SMV often deal with its effect on single or various components of yield studied on single plants or groups of plants to better understand the physiological or economic background of loss in quantity and quality. The data are often hard to compare because of considerable variation according to soybean cultivar, virus strain, and time of infection or inoculation.

Infected plants and their organs are usually smaller in size and weight; plants are often stunted. Reduction in fresh weight of 60-day-old seedlings ranged from 35 to 73% depending on the virus strain (Tu, 1989). In 10 soybean cultivars plant height was reduced by 4.0 to 16.9%, number of pods per plant by 8.4 to 33.4%, pod length by 10 to 25.6%, number of seeds per pod by 6.4 to 15.2%, and total seed weight per plant by 6.6 to 17.8% (Haque et al., 1993). In field experiments, when plants of two cultivars (Hampton 266A and Jackson) were inoculated at the first true-leaf stage, average reduction in plant height was 25% (Demski and Jellum, 1975).

SMV infection usually increases protein content in plants as a whole and often does so with N content. The number and quantity of amino acids may also be changed. The ureide-N content was increased in two plant introduction lines of wild soybean (*Glycine soja*). There is a reduced N-fixation by root nodules of diseased plants. Enzyme activity is often altered. A decrease was found in the activity of catalase and of superoxide dismutase, although an increase of it has also been reported, and an increase in peroxidase activity. These substances may play a role in the resistance of soybean to SMV. Lu and Chen (1992) reported a considerable decrease in photosynthesis in infected plants. Plant maturity was delayed in three experiments in Hampton 266A (Demski and Jellum, 1975) and in a number of other susceptible varieties (Tu, 1989).

SMV decreases seed size, particularly in cases of coinfection with bean pod mosaic virus (Ross, 1968). The oil content of seeds is decreased (Demski and Jellum, 1975). In the oil, the percentage of linoleic and linolenic acid decreased, and stearic and oleic acid content increased. In seeds of cv. Wayne, oil content was reduced following infection from 22.15 to 17.10% and in seeds of cv. Amsoy from 23.76 to 17.6% (Nicolaescu et al., 1977). Infection also reduced germination of seed by 7.4%, 22%, or 52.5% in plants inoculated early during development (Hepperly et al., 1979).

Seed quality is also impaired by increased infection with seedborne fungi. Undesirable seed-coat mottling, though not exclusively caused, but usually substantially increased, by SMV and not correlated with seed transmission of the virus, considerably reduces the grade of the seed and restricts its use. Mere presence of the virus in seed is another factor greatly affecting the quality of seed

if meant for certification as planting material and for commercial traffic

Much of the yield reduction by SMV arises from the increased susceptibility of soybean plants to other viruses caused by infection by SMV. If occurring in or on the seeds, these fungi impair seed quality. When comparing closely related SMV-resistant and susceptible soybean lines submitted to natural infection, the incidence of *Phomopsis* spp. in susceptible lines was much higher than in resistant lines (18.7% versus 4.5%) (Ross, 1977). A decrease in susceptibility to fungi has also been reported in seedborne *Cercospora kikuchii* and *Colletotrichum truncatum* and *Cephalosporium gregatum*, the cause of pith browning.

Most of the information on the effect of SMV on yield derives from measurements of losses caused on inoculated or naturally infected single plants or groups of plants. These plants are compared with healthy ones, which must be kept under identical conditions but free from infection either in the greenhouse or in the field at locations or times of the year with little risk of natural spread of the virus. For SMV, it has also been completed by using a strain of the virus that is not aphid-borne (Goodman and Oard, 1980). The effect on susceptible resistant lines was compared with that on closely related resistant lines (Ross, 1977, 1983; Ren et al., 1979a), or yields after severe infection in the field were compared with yields in a relatively virus-free year (Buzzell, 1983). Such experiments provide insight to yield losses that may occur in practice. They demonstrate the dramatic effect the virus may have, depending on cultivar, virus strain, and conditions.

Figure 1. Mosaic, ruffled leaf margins, and leaf malformation on ‘Bragg’ soybean. Photo courtesy of O.W. Barnett, North Carolina State University.

Symptoms/Signs

Symptoms in sap-inoculated soybean seedlings begin with transient vein clearing in the upper trifoliolate leaves, followed by mosaic (Fig. 1). As the disease progresses, the margins of the affected leaves curl downwards and dark green areas along the veins develop into raised puffs or puckering (Fig. 2) or into a more general leaf curling and rugosity. The primary leaves of plants grown from infected seed may show mottling and downward curling. Infected plants

Figure 1. Leaf curl and puckering. Photo courtesy of Nigel Cattlin (CABI, 2004).

are usually stunted because of shortening of the petioles and internodes, particularly when infected while still young or when infected from the seed. Some cultivars develop progressive necrosis of the petioles and stems, bud necrosis, defoliation, and plant death with some strains resembling the bud blight of soybean caused by Tobacco ringspot virus and Tobacco streak virus (Cho et al., 1977).

The pods are reduced in number and size, some are malformed, glabrous or seedless, and the yield may be considerably reduced. The virus can reduce the size of the seed (Demski and Jellum, 1975), particularly in the case of co-infection with Bean pod mottle virus (Ross, 1968), change its chemical composition, diminish seed viability, seed germination, seedling vigor, and thus the quality of the seed.

There is continuing confusion as to the cause of seed-coat mottling (Fig. 3) which is often associated with SMV infection and considerably increased by it (Kennedy and Cooper, 1966, 1967). However, it may also be caused or increased by other virus infections (e.g. Soybean dwarf virus) and by environmental and genetic factors. It is not correlated with seed transmission of the virus and cannot be used as a parameter of infection by SMV and of seed transmission of the virus.

Figure 3. Seed coat mottling. Photo courtesy of L. Bos (CABI, 2004).

There may also be a reduction in the number of roots and their volume. Bacterial nodulation of the roots is often reduced with a lower leghaemoglobin content and reduced nitrogen fixation.

Known Hosts

Major hosts

Glycine max (soybean)

SMV has a narrow natural host range and a high degree of specificity to its main host, soybean. Cultivated soybean is the major natural host of the virus. Natural infection of common bean (*Phaseolus vulgaris*), in which the virus is also seed transmissible (Castaño and Morales, 1983) may also occur. It has increased in Brazil (Costa et al., 1978) and been demonstrated in New York State, from *P. vulgaris* plants with symptoms closely resembling those of Bean common mosaic virus (Provvidenti et al., 1982). Natural infection was also reported from *Cassia occidentalis* in India (Singh and Gupta, 1996), *Centrosema macrocarpum* in Colombia (Morales et al., 1990), *Lupinus albus* in South Africa (Vroon et al., 1988), *Strophostyles helvola* in Arkansas, U.S. (Kline et al, 1997), and *Vicia faba* in China (Xu et al., 1986a), from the non-legumes *Chrysanthemum frutescens* in Italy and *Passiflora* spp. in Colombia.

The virus can be transmitted mechanically to a limited number of plant species, mainly Fabaceae. Systemic infection was induced in the following: *Cassia occidentalis*, *Crotalaria spectabilis*, *Dolichos falcatus*, *Lablab purpureus*, *Hippocrepis multisiliquosa*, *Indigofera hirsuta*, *Tetragonolobus purpureus*, *Lupinus albus*, *L. angustifolius*, *Macroptilium* (*Phaseolus*) *lathyroides*, *Phaseolus nigricans*, *P. speciosus*, *P. vulgaris*, *Scorpiurus sulcatus*, *Sesbania exaltata*, *Trigonella caerulea*, *T. foenum-graecum* and *Vigna unguiculata* (Quantz, 1961; Galvez, 1963). Local lesions were obtained in *Cyamopsis tetragonoloba*, *Lablab purpureus* and *Macrotyloma uniflorum*. The non-legumes *Chenopodium album* and *C. quinoa* react with local lesions (Quantz, 1961; Galvez, 1963), and symptomless local infections were obtained in *Nicotiana benthamiana* and *N. clevelandii* (Vroon et al., 1988).

The symptoms of SMV in experimental hosts are similar to those of other potyviruses, and natural infection in such species may often have been mistaken for infection by other viruses. Symptomless infection in several species, including the weed *Phaseolus speciosus* (Galvez, 1963), suggests that such species may be a natural source of infection. Pathogenicity of the virus on a number of cultivated plant species, such as *P. vulgaris* (Costa et al., 1978), or of potentially important species, such as *Macroptilium lathyroides*, a possible forage plant in Brazil, indicates the potential significance of the virus for cultivated plant species other than soybean.

Known Distribution

The virus occurs wherever soybean is grown as a crop because of prevalent seed transmission. It should be noted that several literature reports are based on visual observation of symptoms rather than reliable diagnosis of the virus and may be of limited value. Countries that report the presence of the virus include: **Asia:** China, India, Iran, Iraq, Japan, Kazakhstan, Korea, Malaysia, Pakistan, Philippines, Sri Lanka, Thailand, Turkey, **Europe:** Bulgaria, Former Yugoslavia, Gemany, Italy, Moldova, Poland, Portugal, Romania, Russian Federation, Serbia and Montenegro, Sweden, Ukraine, **Africa:** Ethiopia, Morocco, South Africa, Tanzania, Uganda, Zambia, Zimbabwe, **North America:** Canada, the U.S., Jamaica, **South America:** Argentina, Brazil, Chile, Venezuela, **Oceania:** Australia, and New Zealand.

Potential Distribution Within the US

The virus is currently documented as present in Hawaii, Iowa, Mississippi, New York, and Virginia. Its distribution, however, is thought to be wherever soybean is grown.

Survey

Survey for SMV has primarily been based on visual observation of plant symptoms. Visual examination of diseased plants, pods or seeds for symptoms

of SMV, however, is not conclusive evidence that the virus is present.

Key Diagnostics

The biological tests for SMV involve inoculation of a series of conventional test plants for determining the artificial host range or assaying for local lesions. For example, the primary leaves of *Phaseolus vulgaris* on an intact plant or on detached leaves, can be incubated on moist filter paper in a closed Petri dish under artificial light at 30 to 32°C (cultivars include 'Processor' and 'Topcrop) or on *Cyamopsis tetragonoloba*, *Macrotyloma uniflorum* or *Lablab purpureus*.

Serological tests such as ELISA are major means to detect SMV. Various types of ELISA (including monoclonal antibodies) (Hill et al., 1984; Diaco et al., 1985) have been found to be highly sensitive for detecting the virus in individual soybean seeds and seed parts (Lister, 1978; Maury et al., 1985), for detecting infestation in groups of seeds (1 in 160; Lister, 1978), and for the evaluation of virus content in soybean cultivars and selections (Moore et al., 1982). A variant, first immobilizing the virus on a membrane and employing monoclonal antibodies for detection (immunoblotting), has been found helpful for differentiation between, and specific detection of, SMV strains (Hill et al., 1989). Most of the methods for the detection of SMV have been developed for routine seed health testing and seed certification.

Monoclonal antibodies are promising for the differentiation between isolates and strains (Hill et al., 1989, 1994), but a reliable panel specifically detecting all biologically well-identified strains is not available yet. Reverse transcription and polymerase chain reaction (RT-PCR) using specific primers is another modern technique to specifically detect and recognize strains of the virus (Omunyin et al., 1996; Kim et al., 1999). It allowed detection of the virus in a 1:1,000,000 dilution and was found to be 1000 times more sensitive than ELISA (Kim et al., 1999). Dot-blot hybridization using a DNA probe complementary to the 3' noncoding region of the coat protein gene is another test used for specifically detecting SMV (Benscher et al., 1996).

In nature, some 35 viruses have been found in soybean plants (Irwin and Schultz, 1981), and several viruses may occur in mixed infections. Their symptoms may overlap, and symptoms also vary greatly according to virus strain, host cultivar and conditions; therefore, the symptoms are of little diagnostic value. The downward curling of affected leaves of soybean and the occurrence of dark-green areas along the veins develop into raised puffs or puckering are suggestive but do not provide conclusive evidence of infection by SMV. Distinct mosaic symptoms are often absent, and symptom expression can vary according to weather conditions and be masked during periods of high temperature (Irwin and Schultz, 1981). Seed-coat mottling, often associated with and enhanced by the virus, does not prove infection by the virus. SMV is distinguished from non-potyviruses infecting soybeans by the presence of granular inclusion bodies in epidermal strips, viewed by light microscopy, the

presence of 750-nm-long flexuous particles in crude plant sap observed by electron microscopy, or reaction with broad-spectrum potyvirus monoclonal antibodies (e.g. the Agdia PTY-1; Jordan and Hammond, 1991). SMV is distinguished biologically from other potyviruses by its relatively narrow host range (Bos, 1972). The strain of SMV can be determined, and distinguished from Bean common mosaic virus using a set of differential cultivars of soybean (Cho and Goodman, 1979, 1982; Lim, 1985) and of *Phaseolus* (Costa et al., 1978): however, the test is laborious and time-consuming.

Conventional serology using polyclonal antisera to SMV may not sufficiently discriminate the virus from other potyviruses unless the reaction is strong, because serological cross-reaction often occurs between potyviruses. For dependability, present diagnostic and detection methods rely heavily on serological techniques using virus-specific monoclonal antibodies and on PCR and dot-blot hybridization even for recognition of strains.

Other pests

Nematodes

Aphelenchoides besseyi

Scientific Name
Aphelenchoides besseyi Christie

Synonyms:
Aphelenchoides oryzae, Asteroaphelenchoides besseyi

Common Name(s)
White tip of rice nematode, bud nematode, rice leaf nematode, strawberry crimp disease nematode, strawberry summer dwarf nematode, summer crimp nematode, white tip nematode

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Secernentea, **Order:** Tylenchida, **Family:** Aphelenchoididae

Reason for Inclusion in Manual

Pest Description
Females: The body of female *Aphlenchoides besseyi* is slender, straight to slightly arcuate ventrally when relaxed; annules fine, indistinct, about 0.9 µm wide near mid-body. Lip region rounded, unstriated, slightly offset and wider than body at lip base, about half as wide as mid-body; labial framework hexaradiate, lightly sclerotized. Lateral fields about one-fourth as wide as body, with 4 incisures. Anterior part of spear sharply pointed, about 45% of total spear length, posterior part with slight basal swellings which are 1.75 µm across. Median oesophageal bulb oval, with a distinct valvular apparatus slightly behind its

center. Oesophageal glands extending dorsally and subdorsally for 4 to 8 body-widths over intestine. Nerve ring about one body-width behind median oeosphageal bulb (Franklin & Siddiqi, 1972).

Excretory pore usually near anterior edge of nerve ring. Hemizonid 11 to 15 µm behind excretory pore. Vulva transverse, with slightly raised lips. Spermatheca elongate oval (up to 8 times as long as wide when fully distended), usually packed with sperm. Ovary relatively short and not extending to oesophageal glands, with oocytes in 2 to 4 rows. Post-vulva uterine sac narrow, inconspicuous, not containing sperm, 2.5 to 3.5 times anal body width long but less than one-third distance from vulva to anus. Tail conoid, 3.5 to 5 anal body widths long; terminus bearing a mucro of diverse shape with 3 to 4 pointed processes (Franklin & Siddiqi, 1972). L = 0.66 to 0.75 mm; a = 32 to 42 (width = 17 to 22 µm): b = 10.2 to 11.4 (oesophagus = 64 to 68 µm): c = 17 to 21 (tail = 36 to 42 µm); V = 68 to 70 (Christie, 1942).

Males: Male *A. besseyi* are about as numerous as females. The posterior end of body is curved to about 180 degrees in relaxed specimens. Lip region, spear and oesophagus as described for female; tail conoid, with terminal mucro with 2 to 4 pointed processes. First pair of ventrosubmedian papillae adanal, second slightly behind middle of tail and third subterminal. Spicules typical of the genus except that the proximal end lacks a dorsal process (apex) and has only a moderately developed ventral one (rostrum). Testis single, oustretched (Franklin & Siddiqi, 1972). L = 0.54 to 0.62 mm; a = 36 to 39 (width = 14 to 17 µm); b = 8.6 to 8.8 (oesophagus = 63 to 66 µm); c = 15 to 17 (tail = 34 to 37 µm) (Christie, 1942).

Biology and Ecology

In rice, anabiotic *A. besseyi* rapidly become active and are attracted to meristematic areas after sowing. During early growth, the nematode is found in low numbers within the innermost leaf sheath, feeding ectoparasitically around the apical meristem (CABI, 2004). The main stem is frequently more infected than subsequent tillers. A rapid increase in nematode numbers takes place at later tillering and is associated with the reproductive phase of plant growth (Huang et al.,1972).

Nematodes are able to enter spikelets before anthesis, within the boot, and feed ectoparasitically on the ovary, stamens, lodicules and embryo (Huang et al.,1972). However, *A. besseyi* is more abundant on the outer surface of the glumes and enters when these separate at anthesis. As grain filling and maturation proceed, reproduction of the nematode ceases, although the development of J3 to adult continues until the hard dough stage (Huang et al.,1972).

The population of anabiotic nematodes is predominantly adult females. These nematodes coil and aggregate in the glume axis. More nematodes occur in filled grain than in sterile spikelets, and infected grain tends to occur more towards the

middle of the panicle. *A. besseyi* aggregate in the glume axis of maturing grain and slowly desiccate as kernel moisture is lost. They become anabiotic and are able to survive for 8 months to 3 years after harvest (Cralley, 1949; Todd and Atkins, 1958). Survival is enhanced by aggregation and a slow rate of drying (Huang and Huang, 1974), but the number and infectivity of nematodes is reduced as seed age increases.

A. besseyi is amphimictic and males are usually abundant, however, reproduction can be parthenogenetic. The optimum temperature for oviposition and hatching is 30°C. At 30°C, the life cycle is 10 ± 2 days and lengthens significantly at temperatures <20°C (Huang et al., 1972). No development occurs below 13°C.

The principal dispersal method for *A. besseyi* is seed. It can be transmitted in flood water in lowland rice, but the survival of nematodes in water decreases as temperature increases from 20 to 30°C. High seeding rates in infected seed beds facilitate local dispersal (Kobayashi and Sugiyama, 1977).

A. besseyi is a foliar pest of strawberry and may be found between leaves in buds. The nematode has rapid life cycles (2 to 3 weeks) and thrives in moist conditions, which enables them to move over plant surfaces in water films (CABI. 2004)

Pest Importance

A. besseyi is widely distributed in rice production areas because of its dissemination in seed, but its importance varies between regions, countries and localities. Within a locality, the incidence and severity of the disease can change from year to year, and is strongly influenced by cultural practices and local rice types. Infection and damage are generally greater in lowland and deep water systems than in upland environments. However, losses of up to 50% have been reported in upland rice in Brazil (da Silva, 1992).

Figure 1. Chlorotic tips observed on rice infected with *A. besseyi*. Photo courtesy of H. Ferris, University of California, Davis.

Damage in a susceptible cultivar largely depends on the percentage of infested seed sown and the number of *A. besseyi* per infested seed. With few exceptions, the former has rarely been determined despite its importance in governing the number of infection loci in a field. Generally, population densities/seed number or weight are counted. Fukano (1962) determined an economic damage threshold

density (300 live nematodes/100 seed).

In the 1950s, typical figures for susceptible cultivars in the U.S. were 17.5, 4.9 and 6.6% in different years (Atkins and Todd, 1959) and 10 to 30% in Japan (CABI, 2004). Tsay et al. (1998) reported yield losses of 44.9, 34.7 and 24.2% when rice plant infestation rates were 57, 34 and 18%, respectively. *A. besseyi* has been controlled in the U.S. by seed treatment and the use of resistant cultivars and is no longer a major pest of rice. Little is known about its potential impact on soybean production.

A. besseyi is an important pest of strawberry in the U.S., south of Arkansas and Virginia (Brown et al., 1993).

Symptoms

Rice: Plants susceptible to *A. besseyi* can be symptomless, but yield loss only occurs in plants showing some symptoms. During early growth, the most conspicuous symptom is the emergence of the chlorotic tips of new leaves from the leaf sheath. These tips later dry and curl, while the rest of the leaf may appear normal. The young leaves of infected tillers can be speckled with a white splash pattern or have distinct chlorotic areas. Leaf margins may be distorted and wrinkled, but leaf sheaths are symptomless.

The viability of *A. besseyi* infected seed is lowered and germination is delayed, and diseased plants have reduced vigor and height (Todd and Atkins, 1958). Infected panicles are shorter, with fewer spikelets and a smaller proportion of filled grain.

In severe infections, the shortened flagleaf is twisted and can prevent the complete extrusion of the panicle from the boot. The grain is small and distorted (Todd and Atkins, 1958), and the kernel may be discolored and cracked. Infected plants mature late and have sterile panicles borne on tillers produced from high nodes.

Strawberry: *A. besseyi* is a foliar pest of strawberry and may be found between leaves and buds. *Aphelenchoides spp.* cause distortion of the leaves (Fig. 2), which is more noticeable on newly formed leaves after growth resumes in spring in areas of the U.S., south of Virginia, Arkansas, and also in Australia (Brown et al., 1993).

Figure 2. *A. besseyi* symptoms on strawberry. Photo courtesy of EPPO.

Known Hosts

Major hosts

Fragaria ananassa (strawberry) and *Oryza* spp. (rice).

Minor hosts
Allium cepa (onion), *Chrysanthemum morifolium* (chrysanthemum), *Colocasia esculenta* (taro), *Cyperus iria* (rice flatsedge), *Digitaria sanguinalis* (large crabgrass), *Dioscorea* spp. (yam), *Dioscorea trifida* (Indian yam), *Glycine max* (soybean), *Hibiscus* (rosemallows), *Ipomoea batatas* (sweet potato), *Polianthes tuberosa* (tuberose), and *Zea mays* (maize).

Wild hosts:
Oryza breviligulata and *Setaria viridis* (green foxtail)

Known Distribution
A. besseyi is very widely distributed and now occurs in most rice growing areas. Its wide distribution has resulted from dissemination in seed.

Countries reporting the nematode presence include: **Asia:** Afghanistan, Azerbaijan, Bangladesh, Cambodia, China, Republic of Georgia, India, Indonesia, Iran, Japan, Korea, Kyrgyzstan, Laos, Malaysia, Myanmar, Nepal, Pakistan, Philippines, Singapore, Sri Lanka, Tajikistan, Thailand, Turkey, Uzbekistan, Vietnam; **Europe:** Bulgaria, Hungary, Italy, Russian Federation, Slovakia, Ukraine; **Africa:** Benin, Burkina Faso, Burundi, Cameroon, Central African Republic, Chad, Comoros, Congo Democratic Republic, Cote d'Ivoire, Egypt, Gabon, Gambia, Ghana, Guinea, Kenya, Madagascar, Malawi, Mali, Nigeria, Senegal, Sierra Leone, South Africa, Tanzania, Togo, Uganda, Zambia, Zimbabwe; **North America:** Mexico, the U.S.; **Central America:** Cuba, Dominica, Dominican Republic, El Salvador, Guadeloupe, Panama; **South America:** Brazil, Ecuador; **Oceania:** Australia, Cook Islands, Fiji, and Papua New Guinea.

Potential Distribution Within the US
The disease was first noticed in the southern U.S. in 1935 but was initially attributed to a nutrient deficiency (Ou, 1972). Currently the disease is present in rice and strawberry production areas of Arkansas, California, Florida, Hawaii, Louisiana, and Texas. Recent detections in California have sparked the concern of California's rice industry. There is currently no information available on its potential distribution in soybean.

Survey
Different sampling methods are used for *A. besseyi*, depending on the stage of crop growth. During early growth and tillering, *A. besseyi* is found in the base of the culm and between leaf sheaths. For immediate inspection, plant tissue can be teased in water to release the nematodes. This method is suitable for tissue of any potential host species. Plant tissue can be stained before examination; this is particularly useful for detecting low numbers. Alternatively, *A. besseyi* can be extracted from chopped tillers placed on a sieve or directly in water. During the reproductive phase in rice, *A. besseyi* is recovered from spikelets and grain by soaking a known number in water for 24 to 48 hours at 25 to 30°C. Quantitative

extraction from rice requires that the glumes are separated from the kernel but remain in the extract. The percentage of infested seed is a useful parameter, but extracting from individual seeds is time consuming. Better recovery is achieved from hulled grain, but extraction from unhulled grain is sufficient for detection of *A. besseyi* (e.g., for quarantine) from a large seed sample.

One method to detect *A. besseyi* from seeds involves manual dehulling of seeds. The steps include: 1) soak seeds in water for 24 hours, 2) dehull seeds with a scalpel and needle, 3) transfer contents (kernels, hulls and water) to a Baermann funnel or a sieve, and 4) recover and count nematodes after 48 to 72 hours. A second method is the modified Baermann or seive method. The steps include: 1) place seeds over a 10 cm, 40 x 40 mesh steel wire dish in a funnel (12 cm in diameter) and fill with 250 ml water, 2) 1et it stand for at least 48 hours, 3) after incubation, draw approximately 20 ml of water into a test tube through a rubber tube attached to the bottom of the funnel, 4) allow the collected water to stand for 1 hour, 5) pipette out excess water and leave 10 to 15 ml in the test tube, and 6) examine the remaining water for nematodes. A mass extraction method has been described (Hoshino and Togashi, 2002).

Key Diagnostics

A. besseyi could be confused with *Ditylenchus angustus*, the rice stem nematode, particularly under the dissecting microscope. The females and juveniles of the two species are very similar under low-power microscopy, although they can be distinguished by the more experienced nematologist using characters such as head shape and form of the oesophageal bulb. The males are more easily distinguished as the tail shape, spicule shape and presence or abundance of bursa are easier to see. Ideally, identification should be confirmed by a competent taxonomist.

PCR studies using a fragment of ribosomal DNA have also been used to separate *D. angustus* from species of *Aphelenchoides* (Ibrahim et al., 1995).

Allen (1952) provided a key to the four important species related to *A. fragariae*. The key is useful in that it provides characteristics to separate four closely related species. Sanwal (1961) listed 33 species and provided a key.

Ditylenchus africanus

Scientific Name
Ditylenchus africanus Wendt, Swart, Vrain, and Webster

Common Name(s)
Peanut pod nematode

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Secernentea, **Order:** Tylenchida, **Family:** Anguinidae

Reason for Inclusion in Manual

Pest Description
The peanut pod nematode was first discovered in 1987 (De Waele et al., 1989), when it was soaked from hulls and seeds showing symptoms resembling those caused by the fungus *Chalara elegans* (syn. *Thielaviopsis basicola*). It was first identified as *Ditylenchus destructor* (De Waele et al., 1989). Since it did not damage potatoes (De Waele et al., 1991) and thrived at high temperatures around 28 to 30°C (De Waele and Wilken, 1990), it was considered a distinct race from the populations found in Europe and the U.S.

However, a molecular study of comparative taxonomy of some populations of *Ditylenchus* spp. by Wendt (1992) threw doubt on this classification. A subsequent study based on characteristics of morphology and restriction fragment length polymorphisms (RFLPs) of ribosomal DNA described the South African population of *D. destructor* as *Ditylenchus africanus* sp. nov.

The morphometrics of the originally described South African population agreed with those reported for *D. destructor*: 6 to 11 lateral incisures, a rounded tail tip, and a long post-uterine sac relative to the vulva-anus distance (De Waele et al., 1989). De Waele et al. (1989) and Wendt et al. (1995), give detailed descriptions of the adult females and males (larvae not described). The following description

is given by Wendt et al. (1995):

Female: Head flattened, about 1.3 µm high and 6.4 to 7.3 µm wide, not offset from, but narrower than rest of body. SEM shows labial area with pore-like stoma opening surrounded by six outer labial sense organs and two large, medial lips, each with a pair of cephalic sensillae. Outline of labial area and head region hexagonal. Amphidial aperture elliptical, directed towards stomal opening. First head annule discontinuous, caused by position of amphidial apertures. Apart from labial disc, four lip annuli in lip region. Stylet delicate, knobs distinct, separated, sloping backwards; shaft about 60% of total stylet length. Median bulb with crescentic valves. Basal bulb overlapping intestine. Postvulval uterine sac 50 to 143 (79.2± 21) µm long, comprising about 8% of total body length or 37 to 85% of vulva-anus distance and equal to 1.5 to 3.7 times vulval body diameter. Egg measurements: 45 to 60 µm x 20.5 to 33.5 µm. Tail elongate-conoid, tapering in posterior one-third to a finely rounded terminus.

Male: Bursa 33 to 60 (47± 8.6) µm long, leptoderan, covering 48 to 66% of tail length. Spicule arcuate ventrad, slightly cephalated.

Biology and Ecology

The biology of *D. africanus* is very closely related to that of the peanut plant. The nematode apparently survives in the soil on fungi and the roots of peanut, and alternate hosts and weeds in very low numbers until the peanut pegs appear in the soil (around 8 weeks after sowing). Hereafter, the nematode penetrates the immature pod at its connection with the peg, entering the exocarp and moving either longitudinally in this cell layer towards the beak-end of the pod (hence the pod discoloration), or through the mesocarp into the endocarp of the hull (shell) (Venter et al., 1995). It then migrates to the seed micropyle from where it invades the seed testa and embryo (giving rise to the seed symptoms). The nematode has not been found within the cotyledons of the seed (Jones and De Waele, 1990). The nematode reproduces within the pod and seed, and at 28°C the life-cycle from adult to adult is 6 to 7 days (De Waele and Wilken, 1990).

Post harvest, the nematode is able to survive in planting seed, which may be symptomless and in the field soil in the absence of host plants or in hulls buried in the soil, for at least 28 to 32 weeks (Basson et al., 1993). This is long enough to survive the dry winter season in South Africa. The nematode primarily survives through a process known as anhydrobiosis, inactivity of the nematode (as indicated by coiled morphology) occurring in response to desiccation of the soil habitat. As physiological maturity approaches (from 17 to 21 weeks after sowing, varying between cultivars) the relative number of eggs and anhydrobiotes in the pod and seed tissues increases (Venter et al., 1995).

With the first spring rains the eggs hatch and anhydrobiotes rehydrate. Basson et al. (1993) showed that rehydrated soil populations of *D. africanus* are able to invade the next crop of peanut and cause damage. Although relatively few nematodes survive in whole stored seed at 10°C, the surviving nematode

population is also able to build up to levels capable of causing damage to the next crop (Basson et al., 1993).

Pest Importance

Although *D. africanus* is suspected of being present in many southern African countries, it has only been reported to cause damage in South Africa. In this country, it is found in all the major peanut production areas, on about 75% of all fields (De Waele et al., 1989).

It is a pest of the pods of peanuts on which it can multiply to heavy infections (100,000 nematodes/pod), causing 100% losses in some fields. It is found only in low numbers on the roots of peanuts or alternate crops (Basson et al., 1990), and causes no damage to the tubers of potatoes (De Waele et al., 1991), which is the host of the closely related *D. destructor*.

A 1987 survey of damaged seeds obtained from 877 farms representative of the major peanut production areas, showed that 73% of the samples were infected with *D. africanus* (De Waele et al., 1989). The greatest economic damage of *D. africanus* is the increase in the percentage of seeds which are blemished (discolored) and/or unsound (germinating seeds within closed pods; hypocotyl 1 to 2 mm) (Venter et al., 1991). In heavy infections (final density in excess of 700 nematodes/seed) the seed mass may be reduced by up to 50%, and untimely germination of seedlings may reduce the number of harvestable seeds by up to 25% (Venter et al., 1991). This is a major factor in determining the grade of the yield. South African grading regulations require that consignments of peanut seed containing more than 10 and 20% damaged seed be downgraded from choice edible to standard edible and to crushing grade, respectively, resulting in price decreases of 15 and 65%, respectively.

Figure 1. (Left) Symptoms of *D. africanus* on peanut hulls and seeds. (Right) Symptoms of peanut black hull caused by *Theilaviopsis basicola* (*Chalara elegans*). Photos courtesy of CABI, 2004 and J. Damicone, Oklahoma State University.

Symptoms

D. africanus does not cause visible lesions on the roots of peanuts, but affects the appearance, and sometimes the weight and untimely germination, of the pods and seeds (Venter et al., 1991). Infected hulls also show greyish-black to brown necrotic tissue at the point of connection with the peg, and in broad bands along the longitudinal veins (Fig. 1). Infected seeds are usually shrunken with dark brown to black micropyles and yellow to dark flaccid testae with dark vascular strands. The embryos may also become darkly discolored (Jones and De Waele, 1990).

No symptoms are visible on the roots of alternate hosts (Basson et al., 1990) or the tubers of potatoes (De Waele et al., 1991).

Known Hosts

Major host

Arachis hypogea (peanut)

Minor hosts

Chenopodium album (fat hen), *Datura stramonium* (jimsonweed), *Eleusine indica* (goose grass), *Glycine max* (soybean), *Gossypium hirsutum* (Bourbon cotton), *Helianthus annuus* (sunflower), *Lupinus albus* (white lupine), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Tagetes minuta* (stinking Roger), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), *Xanthium strumarium* (common cocklebur), and Zea mays (maize)

Fungal hosts

Chalara spp., *Penicillium* spp., *Phytophthora* spp., *Aspergillus* spp. and *Fusarium* spp. (CABI, 2004).

Known Distribution

Mozambique and South Africa

Potential Distribution Within The US

No information is available at this time

Survey

D. africanus is extremely difficult to extract from soil or from the roots of any crops. The quickest detection, in the field, is by inspection of the mature pods for the characteristic grey discoloration, which begins at the peg connection and develops in broad bands down one or both longitudinal veins, until the entire pod surface is discolored (Venter et al., 1991). The inside of the shell is also discolored and the seed testae flaccid and darkly discolored. If in doubt, the shells and seeds can be soaked in tap water for 24 hours (Bolton et al., 1990) and the water inspected for the presence of *D. africanus:*

1. A subsample of peanut seeds is taken and the seed cut open.

2. The cut seeds are soaked in tap water for 24 hours at approximately 22°C.

3. The nematodes in the water are poured off and counted.

The efficiency of the soaking method was significantly higher (2 x for hulls; 3 x for seeds) and more consistent (as expressed by the coefficient of variation) than the efficiency of the centrifugal flotation method. The soaking method is also an inexpensive and rapid method involving few steps. The absence of any sieving during the soaking method may have reduced the loss of larvae compared with the centrifugal-flotation method. The recovery of immobile adults by the soaking method indicates that the nematodes not only actively moved out of the tissues but that they were passively released by swelling and bursting of the tissues (Bolton et al., 1990). The total number of nematodes recovered by soaking after 14 days appears to be a good estimation of the total number of all life stages, including eggs, present inside the tissues at the beginning of the soaking period. Soaking for 24 hours (x) gives a reliable estimate of this number (y): y = 37,415 + 1132 x (r = 0.911; P = 0.05) for hulls, and y = 48,663 + 1411 x (r = 0.827; P = 0.05) for seeds (Bolton et al., 1990). Stored seeds release far fewer nematodes when soaked. These are most often dead, and their diagnostic features are in a deteriorated condition (CABI, 2004)

Key Diagnostics

In South Africa, peanut producers often confuse the symptoms of *D. africanus* with those of the fungus *Chalara elegans* (Fig. 1). Symptoms are distinguished by the pattern of development of pod discoloration. That of *D. africanus* begins at the peg connection, develops in broad bands down one or both lateral veins, before covering the entire pod surface (Venter et al., 1991). It causes a grey discoloration of the mesocarp, which cannot be removed by scratching off the exocarp of the pod, and is visible on the inside of the shell. That of *C. elegan*s begins at any and various points on the pod, is dark black, and can be scratched off with the exocarp. The seeds of pods infected by *D. africanus* have yellow to brown flaccid testae, often with darkened veins, and discolored embryos; while those infected by *C. elegans* are not discolored by the fungus (CABI, 2004).

D. africanus is very similar morphologically to *D. myceliophagous* with respect to the number of lines in the lateral field, shape of the tail terminus, c', c, stylet length, length of the post-uterine sac expressed in vulval body diameters, V-value, and spicule and bursa length. However, it differs significantly from *D. myceliophagous* in its molecular character and host specificity (Wendt et al., 1995). *D. africanus* is also very similar to *D. destructor* (Wendt et al., 1995). De Waele et al. (1989) first considered it to be a race of *D. destructor* with a limited host range. These three species can only be clearly differentiated using sensitive RFLP analysis (Wendt et al., 1995).

Although PCR was also used by Wendt et al. (1993) to distinguish *D. dipsaci* from *D. destructor* and *D. myceliophagus*, it is only one of several tools required for identification of these species. A description of morphology and preferably also of host plants and ecology would be required to complete the species identification.

Ditylenchus destructor

Scientific Name
Ditylenchus destructor Thorne

Common Name(s)
Potato tuber nematode, potato eelworm

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Secernentea, **Order:** Tylenchida, **Family:** Anguinidae

Reason for Inclusion in Manual

Pest Description
Thorne (1945) proposed and described *Ditylenchus destructor* from potato in Aberdeen, Idaho. Before *D. destructor* was described in 1945 as a new species, it was regarded for a long time as a strain or race of *D. dipsaci.* Much of the earlier literature, therefore, provides confused information on the two species, especially in relation to potatoes. *D. destructor* can easily be differentiated from *D. dipsaci* in having six incisures in the lateral field (as against four) and a rounded tail terminus (pointed in *D. dipsaci*).

There is considerable morphological variation shown by the adults of *D. destructor* due to age or feeding on particular hosts. Body slender (a=30 to 35 µm). Cuticle smooth, marked by faint and fine transverse striae about 1 µm apart; lateral field with six incisures. Cephalic region smooth, low, anteriorly flattened, slightly set off or almost continuous with body contour. Cephalic framework hexa-radiate, moderately developed. Stylet slender, 10 to 14 µm long, with distinct basal knobs. Median oesophageal bulb fusiform; basal bulb clavate, usually its base overlaps the intestine on the dorsal side for half to one body width. Excretory pore at or just anterior to oesophago-intestinal junction; hemizonid just in front of excretory pore. Tail conoid, slightly arcuate ventrally, with a minutely rounded tip.

According to Thorne (1945), female length ranges from 0.81 to 1.4 mm (a=30 to 35 µm; b=8 to 10 µm; c=15 to 20 µm; V=78 to 83%). Vulva a transverse slit, at 78 to 83% of body length from anterior end. Ovary single, outstretched anteriorly, sometimes reaching the oesophagus; oocytes in double rows in anterior region, then in single file. Spermatheca elongate-oval, often with large sperm arranged in a row. Post-vulval uterine sac about 75% of vulva-anus distance. Tail 3 to 5 anal body widths long, with a minutely rounded tip.

Male length ranges from 0.8 to 1.3 mm (a=34 to 40 µm; b=7 to 8 µm; c=12 to 16 µm; T=73 to 80%. Males are abundant and similar to female in general appearance. Testis single, outstretched; sperm large-sized, rounded, in 1 to 2 rows. Spicules large and prominent, ventrally arcuate. Gubernaculum linear. Bursa enveloping about four-fifths of tail.

There are four juvenile stages, resembling female in general morphology but lacking genital structures, The first stage occurs within the egg, which is oval, about twice as long as wide.

Biology and Ecology
D. destructor is a migratory endoparasite of roots and underground modified plant parts such as potato tubers, bulbous iris and garlic. The nematodes attack the subterranean and only rarely the aerial parts of plants. They enter potato tubers through the lenticels, and then begin to multiply rapidly and invade the whole tuber. They can continue to live and develop within harvested tubers.

D. destructor attacked carrots at the base of the lateral roots, and tissue breakdown occurred in the cortex. The damaged tissue was discolored. External lesions subsequently appeared, which served as infection sites for other pathogens, including *Mycocentrospora acerina*. It was found to attack stems, buds and leaves of *Cimicifuga racemosa* (Planer, 1972) and roots of ginseng in Korea (Young and Seung, 1995). Stem infestations are rare but have also been reported on potato haulm by Goodey (1951) and on *Vicia sativa* by Duggan and Moore (1962).

The nematodes can move only short distances in the soil and have no natural means of long-range movement. The main means of dispersal is with infested potato tubers or other subterranean organs of host plants, for example bulbs and rhizomes. Transport in infested soil is another important means of spread. Irrigation water can also carry the nematodes. Unlike the closely related species *D. dipsaci*, *D. destructor* is unable to withstand excessive desiccation, and for this reason is usually important only in cool, moist soils. Unlike *D. dipsac*i, it does not form 'eelworm wool'. Without a resistant resting stage, the species overwinters in soil as adults or larvae and may even multiply by feeding on alternative weed hosts and on fungal mycelia. It may also possibly overwinter as eggs. These hatch in the spring and larvae are immediately able to parasitize

hosts. Thorne (1961) suggested that *D. destructor* overwintered in U.S. field soil as eggs and coiled adults.

There appears to be a synergistic relationship between *D. destuctor* and *Rhizoctonia solani* and *Fusarium* spp. in potato tubers. Infection by *R. solani* was highest in pots to which the largest number of *D. destructor* (136 nematodes per 100 g soil) was added. The results confirmed that mixed infections were more harmful to the potatoes than either infection alone (Janowicz and Mazurkiewicz, 1982). The damage to potato tubers stored in the dark at 6 to 15°C was greater (49% compared to 27%) when both the dry rot Fusaria (*Fusarium solani* var. *coeruleum*, *F. culmorum* and *F. oxysporum*) and *D. destructor* were present, than when only the dry rot fungi were present (Janowicz, 1984).

Pest Importance

In general, *D. destructor* can become important as a pest of potatoes at temperatures of 15 to 20°C and at relative humidity above 90%. Healthy seed potatoes planted in infested fields in Sweden resulted in crops damaged by 0.3 to 94%; severely infested seed tubers exhibited external symptoms in 41 to 70% by weight of the new tubers (Andersson, 1971). The degree of infestation of potato tubers by *D. destructor* on Estonian farms ranged from 2 to 9%. Up to 80 to 90% of tubers from some fields became infected during storage (Kikas, 1969).

Figure 1. External symptoms, showing sunken areas with cracked and wrinkled skin. Photo courtesy of EPPO. 1712Hhttp://www.eppo.org/QUARANTINE/nematodes/Ditylenchus_destructor/DITYDE_images.htm

When animals were fed potato tubers infected with *D. destructor* or were injected with extracts from such tubers, the intensity of antibody production was reduced by half or more, and the phagocytic activity of leukocytes and the cholesterol content of the blood were also reduced (CABI, 2004).

Symptoms

There are, in general, no obvious symptoms in the aerial parts of the potato plant, although heavily infested tubers give rise to weak plants which usually die. Early infections can be detected by peeling the tuber, which can reveal small, off-white spots in the otherwise healthy flesh. These later enlarge, darken, are woolly in texture and may be slightly hollow at the center. If stored in moist conditions, a general rot may ensue and spread to other tubers. Infested dahlia tubers develop similar symptoms (CABI, 2004; Hooper, 1973).

On badly affected tubers, there are typically slightly sunken areas with cracked and wrinkled skin (Fig. 1), which is detached in places from the underlying flesh. The flesh has a dry and mealy appearance, varying in color from greyish to dark brown or black (Fig. 2). This discoloration is largely due to secondary invasion of fungi, bacteria and free-living nematodes (the latter are easily confused with *D. destructor*). In contrast, the skin of potatoes infested with *D. dipsaci* is not usually cracked, and the rot darkens towards the inside of the tuber. The symptoms are more obvious in the foliage, which is shortened and malformed. Rotting due to *D. destructor* in storage increased with rising temperature, but there was no evidence of transfer of infestation from diseased to healthy tubers (Andersson, 1971).

Figure 2. Potato infected with *D. destructor* (right) compared with a noninfected one (left). Photo courtesy S. Ayoub.

On iris and tulips, infestations usually begin at the base and extend up the fleshy scales, causing grey-to-black lesions; roots may be blackened, and leaves poorly developed with yellow tips.

Figure 3. Damage to Iris bulbs ranging from clean to severe damage. Photo courtesy of EPPO.
1713Hhttp://www.eppo.org/QUARANTINE/nematodes/Ditylenchus_destructor/DITYDE_i

Known Hosts

Potato, sweet potato and bulbous iris are the main hosts of *D. destructor*; occasionally tulips, gladioli and dahlias become important hosts. Root crops sometimes affected include sugar beet, mangolds (*Beta vulgaris)* and carrots. Clovers (*Trifolium* spp.), cultivated mushrooms, onion and garlic are also good hosts. Hooper (1973) states that some 70 crops and weeds and a similar number of fungus species have been recorded as hosts (Goodey et al., 1965).

Major hosts

Allium cepa (onion), *Allium sativum* (garlic), *Arachis hypogaea* (peanut), *Beta vulgaris* (beet), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Camellia sinensis* (tea), *Capsicum annuum* (bell pepper), *Chrysanthemum morifolium* (chrysanthemum), *Citrus sinensis* (navel orange), *Cucumis sativus* (cucumber), *Cucurbita moschata* (pumpkin), *Dahlia* hybrids, *Daucus carota* (carrot), *Fragaria*

ananassa (strawberry), *Gladiolus* hybrids (gladiolas), *Glycine max* (soybean), *Humulus lupulus* (hop), *Ipomoea batatas* (sweet potato), *Iris* (irises), *Lycopersicon esculentum* (tomato), *Mentha* (mints), *Panax ginseng* (Asiatic ginseng), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Trifolium* (clovers), *Triticum aestivum* (wheat), *Tulipa* (tulip), and *Zea mays* (maize)

Wild hosts
Chenopodium album (fat hen), *Cyperus rotundus* (purple nutsedge), *Datura stramonium* (jimsonweed), *Eleusine indica* (goose grass), *Elymus repens* (quackgrass), *Fumaria officinalis* (common fumitory), *Solanum* (nightshade), *Sonchus arvensis* (perennial sowthistle), *Tagetes minuta* (stinking Roger), *Taraxacum officinale* complex (dandelion), and *Xanthium strumarium* (common cocklebur)

D. destructor can feed on fungi, and can easily be cultured on many fungi and on plant callus (Darling et al., 1957; Faulkner and Darling, 1961). It is readily established on laboratory cultures of *Alternaria tenuis* (*A. alternate*) and *A. solani* (Foot and Wood, 1982). *D. destructor* reproduced well on cultures of *A. tenuis* on potato glucose agar at 26 to 27°C. It was also cultured on ginseng root callus, fungal mycelium (*Fusarium solani*), carrot discs and radish sprouts (Young and Seung, 1995).

Known Distribution
D. destructor is a pest of potatoes mainly in temperate regions: localized areas in North America, many parts of Europe, the mediterranean region, and Asia.

Pre-1995 records from peanut and several weeds in South Africa are now considered to be of *D. africanus,* although *D. destructor* has recently been validly reported from this country (CABI, 2004).

Countries with the nematode present include: **Asia:** Azerbajan, China, Iran, Japan, Kazakhstan, Korea, Kyrgyzstan, Pakistan, Saudi Arabia, Tajikistan, Turkey, Uzbekistan; Europe: Albania, Austria, Belarus, Belgium, Bulgaria, Czech Republic, Estonia, France, Germany, Greece, Hungary, Ireland, Latvia, Lithuania, Luxemburg, Moldova, Netherlands, Norway, Poland, Romania, Russian Federation, Slovakia, Sweden, Switzerland, Ukraine, United Kingdom; **Africa:** South Africa; **North America:** Mexico, the U.S.; **South America:** Ecuador, Peru; **Oceania:** Australia, and New Zealand.

Potential Distribution Within the US
The nematode has been reported in: Arkansas, California, Hawaii, Idaho, Indiana, New Jersey, North Carolina, Oregon, South Carolina, Virginia, Washington, West Virginia, and Wisconsin. In Wisconsin, the spread of the pest has been stopped through the elimination of infection sources by fumigation, a strict state quarantine limiting movement of infected tubers, and supervision of the disposition of potatoes from infested fields (Darling et al., 1983).

The requirement of the nematode for high relative humidity means it would be unlikely to become a problem in areas with warm, dry soils. It may, therefore. be of concern to potato production only in the cooler areas.

Survey

Prior to planting, soil can be sampled using a standard extraction procedure for nematodes of this size (Hooper, 1986). Microscopic examination of the nematode is necessary for correct identification of the species.

It is difficult to detect the presence of *D. destruct*or on potatoes from external tuber appearance alone. Sample tubers should be cut or peeled to look for the characteristic whitish pockets, in which most of the nematodes are found. However, on badly affected potato tubers there are typically slightly sunken areas with cracked and wrinkled skin which is detached in places from the underlying flesh. The flesh has a dry and mealy appearance, varying in color from greyish to dark brown or black. External symptoms on iris and tulip include grey-to-black lesions; heavily infested bulbs often have blackened roots and poorly developed, yellow-tipped leaves.

Key Diagnostics

The *Ditylenchus* species that attacks peanut in South Africa has been referred to as *D. destructor* in the literature but is now considered a different species, *D. africanus* (Wendt et al., 1995). *D. destructor* was referred to as a race o*f D. dipsaci*. *D. destructor*, however, can be easily be differentiated from *D. dipsaci* by the six incisures in the lateral field (as compared to four) and a rounded tail terminus (pointed in *D. dipsaci*).

A PCR based technique was also used by Wendt et al. (1994) to distinguish *D. dipsaci* from *D. destructor* and *D. myceliophagus*. However, it is only one of several tools required for identification of these species. A description of morphology and preferably also of host plants and ecology would be required to complete the species identification.

Heterodera glycines

Scientific Name
Heterodera glycines Ichinohe

Common Name(s)
Soybean cyst nematode

Type of Pest
Nematode

Taxonomic Position
Class: Secernentea, **Order:** Tylenchida, **Family:** Heteroderidae

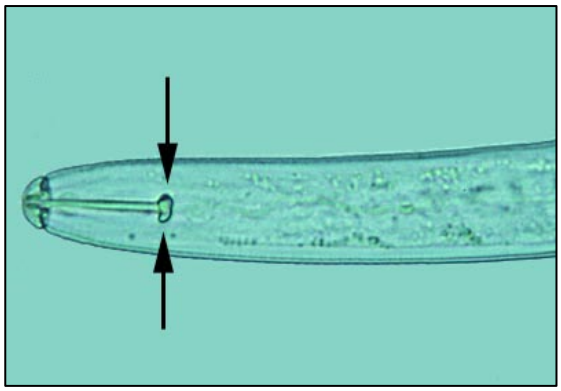


Figure 1. Vermiform (worm-shaped) nematode with stylet. Note arrows point to stylet knob. Photo courtesy of 1695Hwww.apsnet.org

Reason for Inclusion in Manual

Pest Description
Nematodes are unsegmented roundworms. Most plant parasitic types are very small and feed on roots by means of a stylet (Fig. 1), a hollow, needle-like structure used to pierce plant cells and withdraw nutrients.

Soybean cyst nematode (SCN), *Heterodera glycines*, was first formerly described by Ichinohe from soybean in Hokkaido, Japan in 1952. The nematode was first discovered in the U.S. at Castle Hayne, North Carolina in 1954. Currently, SCN is one of the most serious pests of soybean and has spread to 28 soybean producing states and Canada. Some areas of the U.S. are so heavily infested, that soybean production is no longer economically possible without control measures.

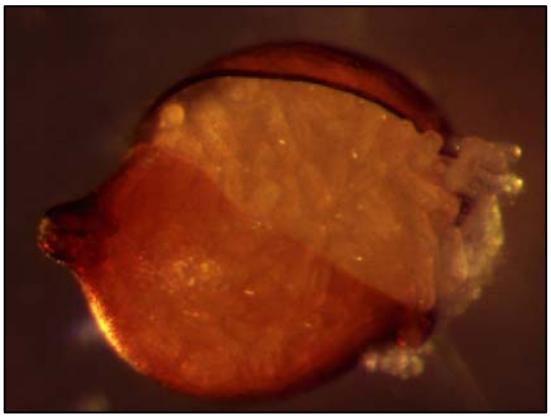


Figure 2. Cyst broken open to reveal numerous eggs contained inside. Photo courtesy of Nemapix (McGawley).

The mature female is an obese, sedentary semi-endoparasite of plant

roots. Vermiform adult males may be found in the soil. The eggs are normally retained in a cyst formed from the cuticle of the dead female (Fig. 2).

Female: Morphology is typical of the genus. Body swollen, lemon-shaped with projecting neck containing the oesophagus and part of the oesophageal glands (Fig. 3). Body without annulation or lateral incisures but covered with reticulate ridges. Females white on emergence from the root cortex, turning pale yellow as eggs develop. Gelatinous matrix or egg sac is present containing up to 200 eggs. Sub-crystalline layer prominent. Head skeleton hexaradiate, stylet slender with posteriorly projecting knobs. Median bulb large and subspherical. Vulva and anus carried on an obtuse cone-shaped projection opposite the neck. Vulva a transverse slit on the vulval cone terminus, surrounded dorsally and ventrally by thin walled crescent-shaped areas, the semifenestrae (Ichinohe, 1955; Hirschmann, 1956; and Burrows and Stone, 1985). On death, the female body wall tans to form a brown, tough walled cyst containing hundreds of eggs (Fig. 2, 4).

Figure 3. Young white female. Photo courtesy of Anne-Sophie Roy 1696Hwww.invasive.org

Figure 4. Crushed SCN female with eggs. Photo courtesy of Plant & Pest Diagnostic Lab, Purdue University.

Cyst: Lemon shaped with protruding neck and vulval cone (Fig. 2). Outer cyst wall with a rugose pattern of zigzag lines (Fig. 5). Ambifenestrate. Vulval region may be intact on younger cysts, but in older specimens the thin walled cuticle of the terminal region is lost leaving an open fenestra crossed by the vulval bridge bearing the vulval slit and dividing the fenestra into two semifenestrae. Bullae prominent, elongate, at or just below the level of the well-developed underbridge. Japanese population: L = 700 ± 60 µm; maximum width = 490 ± 54 µm; length/width = 1.43 (1.20 to 1.61). USA population: L =

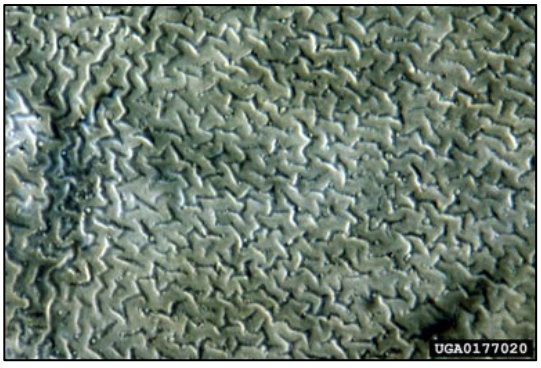


Figure 5. Cyst wall with typical zig-zag pattern. Photo courtesy of Anne-Sophie Roy. 1697Hwww.invasive.org

340 to 920 µm; length/width = 1.19 to 2.05; vulval slit = 49.7 (43 to 56) µm; fenestral length = 53.7 (37 to 65) µm; fenestral width = 40.5 (33 to 48) µm (Ichinohe, 1955; Hirschmann, 1956; and Burrows and Stone, 1985).

Male: Vermiform with short, bluntly rounded tail region. Cuticle regularly annulated. Lateral field with four incisures. Head offset with 4 to 5 annules and strong head skeleton. Stylet robust with knobs laterally to anteriorly projecting.

Figure 6. Head (left) and tail (right) of vermiform juvenile *H. glycines*. Note tail has a hyaline terminus. Photo courtesy of Anne-Sophie Roy. 1698Hwww.invasive.org

Dorsal oesophageal gland opening 4 µm posterior to stylet base. Excretory pore funnel shaped and 14.5 µm from head. Dorsal oesophageal gland lobe overlapping intestine ventrally. Spicules strongly developed; gubernaculum present (Ichinohe, 1955; Hirschmann, 1956; Burrows and Stone, 1985).

Second stage juvenile: Vermiform with four incisures in the lateral field, the incisures reducing to three anteriorly and posteriorly. Head offset with 3 or 4 annules. Labial disc dumb-bell shaped. Stylet robust with anteriorly directed knobs. Anterior and posterior cephalids located 2nd to 3rd and 7th to 9th annules respectively. Tail tapering uniformly to a finely rounded terminus; hyaline portion about 50% of tail length (Fig. 6). Japanese population: L = 471 (437 to 504) µm; width = 18.3 (18.0 to 18.5); stylet = 23.1 µm; tail length = 45.0 (42 to 47) µm. USA population: L = 440 (375 to 490) µm; stylet length = 23.0 (22.0 to 24.0) µm; tail length = 50.4 (42.0 to 59.4) µm; length hyaline tail terminus = 26.6 (20.0 to 33.0) µm (Ichinohe, 1955; Hirschmann, 1956; Burrows and Stone, 1985).

Figure 7. Second stage juvenile inside egg. Photo courtesy of Anne-Sophie Roy. 1699Hwww.invasive.org

Biology and Ecology

The lifecycle of SCN is similar to other cyst nematodes. Six stages are involved in the life cycle of this nematode: an egg stage, four juvenile stages designated J1 to J4, and the adult stage. The nematode reproduces amphimictically. Adult females are lemon-shaped and are semi-endoparasites of plant roots. After death, the cuticle tans to form a brown cyst that serves to protect the retained eggs, although numerous eggs are laid in an external gelatinous matrix. Females may produce up to 600 eggs each, 200 of which may be in an egg sac. Eggs remain viable in the cyst for up to 11 years. The first stage juvenile (J1) undergoes the first molt while still inside the egg and the second stage juvenile (J2) hatches from the egg (Fig. 7). The J2, the infective stage, seeks a host root and penetrates the cortex (Fig. 8). The vermiform nematode then becomes sedentary and feeds via specialized trophic cells formed by the host in response to secretions from the nematode. The developing nematodes become increasingly obese and molt to the J3. Ultimately, the J4 stage is reached. The J4 molts to the female, which remains in position within the root cortex or molts to the vermiform male which escapes from the J4 cuticle and the root and searches for females to mate with. The nematode may complete several generations per year.

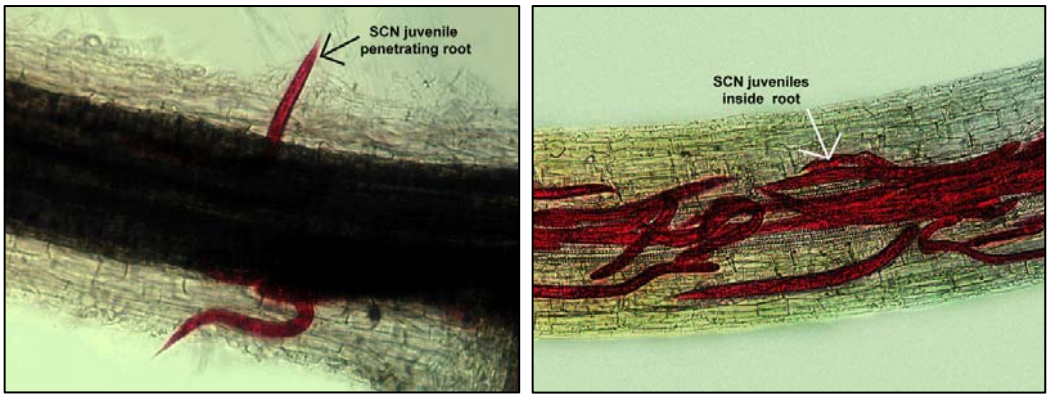


Figure 8. SCN juveniles penetrating root and inside root. Photo courtesy of Nemapix (Eisenback).

The SCN development is directly dependent on soil temperature and moisture. At soil temperatures of 21 to 23°C and with sufficient moisture, the lifecycle requires 21 to 24 days. At cooler temperatures, 18°C, the life cycle requires approximately 40 days. Optimum temperature and development is 28 to 31°C, little development occurs at temperatures below 15°C or above 33°C. The optimum temperature for emergence of the J2 from the egg and for root penetration is reported to be 24°C (CABI, 2004).

Cyst population density was consistently higher in loamy sand than in sandy clay loam.

SCN field populations vary in their abilities to successfully develop and reproduce on a set of four differential soybean lines that differ genetically in their resistance to SCN. These different populations are referred to as SCN races and are given number designations. There are currently 16 possible reaction combinations, and thus, 16 potential SCN races. To date 12 different races have been reported in the U.S. Such a differential response to cultivars serves to complicate management strategies involving resistant hosts, particularly if more than one nematode race is present (CABI, 2004).

Pest Importance

Due to its severe injury to host plant roots, rapid reproduction, and persistence in the soil, SCN is considered a serious agricultural pest. The lifecycle of SCN is completed in about one month. It is possible to have three to six generations in a single cropping year, depending upon location. SCN enters the root tissue of susceptible plants and feeds internally.

SCN is the single most damaging pest of soybeans in the U.S. SCN may decrease yields substantially without inducing obvious symptoms. The resilience of SCN makes management and not eradication the most viable option for minimizing its impacts on soybean production. Consequently, many fields are infested without the knowledge of the grower. Over the last 25 years, SCN has moved increasingly northward from the southern U.S. and is now damaging tens of millions of soybean acres. One estimate of the soybean crop loss in north central states attributable to *H. glycines* parasitism was over 200 million annually (Doupnik, 1993).

Figure 9. Closeup of soybean stunted from infection by soybean cyst nematode (right) compared with healthy plant (left). Photo courtesy of Nemapix (Eisenback).

Reported yield losses on soybean vary from 10 to 70% in Japan. All soybean growing areas in the U.S. are at risk, and the nematode is still spreading into previously uninfested areas. Losses in the southeastern U.S. were estimated at $88.4 million in 1990 (Sciumbato, 1991). Wrather et al. (1997) provided loss estimates for the top 10 soybean producing countries and concluded that, worldwide, *H. glycines* was the most important constraint on yield.

The nematode presents a threat to all regions of the world where soybeans are grown. Steps should be taken to prevent introduction in the first instance and to

control spread once the nematode is known to be present. The nematode is spread most easily via infested soil and contaminated machinery. Any mechanism that spreads infested soil can be a means of dispersal, including wind, water, migratory birds, and pods in seed lots. *H. glycines* is already widespread in most of the countries where soybean production is a major agricultural activity. SCN can only move a few inches on its own, but despite federal quarantines, it spread from a localized infestation in 1954 to 65 counties in eight soybean producing states by 1965, to 15 states in 1976, to 22 states in 1980, and to a total of 540 counties in 24 states by 1985 (PPQ, 1993).

Symptoms/Signs

The presence of SCN is not usually obvious at the time of initial soil infestation. To be detected, the nematode population density must increase in the soil until it is sufficient to cause above-ground symptoms on plants or a decrease in yield. Population densities may take several years to reach significant numbers. Thus, current SCN damage is a result of infestations that have been growing for several years.

Figure 10. Resistant and susceptible soybeans growing in a *Heterodera glycine*s infested field. Photo courtesy of Nemapix (J. P. Ross).

'Yellow dwarf' is an appropriate description for symptoms that are commonly caused by SCN. When soybean plants are severely infected, they become stunted (Fig. 9), canopy closure does not occur, and leaves may become chlorotic (Fig. 10). Unfortunately, these symptoms are not unique to the disease caused by SCN and may be confused with symptoms caused by other crop stresses such as iron deficiencies, injury from agricultural chemicals, feeding

Figure 11. Aerial view of SCN damage. Photo courtesy of Greg Tylka, Iowa State University.

of soybean aphid, and infection by other plant pathogens. The first obvious symptom of SCN injury is the appearance in the field of circular- or oval-shaped areas of stunted, yellowed, less vigorous plants (Fig. 11). These infested areas will vary in their size, often showing a sharp dividing line at the edges between stunted and apparently healthy plants. Plants growing in heavily infested soils may remain stunted throughout the season. Rows of soybean grown on SCN-infested land frequently are slow to close or fill in with foliage.

There are differences between symptoms of SCN and iron deficiency chlorosis. Iron deficiency chlorosis symptoms usually appear in early June, whereas yellowing due to SCN will most likely occur in July and August. The yellowing caused by iron deficiency chlorosis typically affects the areas between the veins of the upper leaves. Yellowing due to SCN usually starts at the edges of the leaves and can affect leaves on the entire plant. Iron deficiency chlorosis and SCN may occur in the same field and even on the same plant.

One cannot rely upon above-ground symptoms for identification of SCN infestations. If soybean yields in any field have decreased for no apparent reason, or if SCN has been confirmed on nearby land, more thorough examination of plants for below-ground symptoms and a soil analysis for SCN are needed.

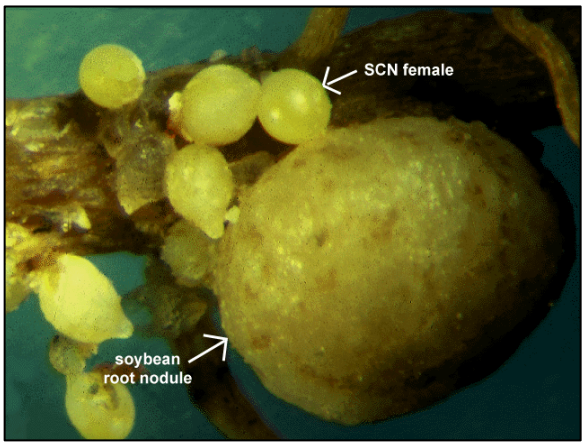


Figure 12. Female soybean cyst nematodes, *H. glycines*, compared to a nodule on the root system. Photo courtesy of Nemapix (McGawley).

Most below-ground symptoms of SCN damage are not unique. Roots infected with SCN are dwarfed or stunted. SCN can decrease the number of nitrogen-fixing nodules on the roots. SCN infections also make roots more susceptible to attack by other soilborne plant pathogens. Often, it is difficult to recognize if roots are stunted and have fewer nodules unless they are compared with uninfected soybean plants. The only unique characteristic of SCN infection is the presence of adult female and cysts on the soybean roots. These structures, which appear as tiny, lemon-shaped objects on the roots, are white initially, but turn yellow and then tan to brown as they mature. They can be seen with the unaided eye, although observation with a magnifying glass is easier. The cysts are about the size of a pinhead and much smaller than nitrogen nodules (Fig. 12). Cysts can be observed from 4 to 5 weeks after planting, are usually abundant in July and August, and then decline in numbers as the roots senesce. Roots must be carefully removed from the soil for examination or the cysts may

be dislodged. Observation of adult females and cysts on the roots of soybean plants is the only accurate way to diagnose SCN infestation in the field.

Known Hosts

H. glycines attacks a wide range of Fabaceae. Members of Carophyllaceae and Scrophulariaceae are also hosts. Riggs and Wrather (1992) give a list of non-fabaceaous hosts comprising 63 species in 50 genera from 22 families. The soybean cyst nematode infects soybean, dry edible bean, and snap bean, but not rotation crops such as corn, small grains, and alfalfa.

Major Host

Glycine max (soybean)

Minor Hosts:

Cajanus cajan (pigeon pea), *Glycine* spp., *Kummerowia striata* (Japanese lespedeza), *Lespedeza cuneata* (Sericea lespedeza), *Lupinus* (lupine), *Lycopersicon esculentum* (tomato), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Vicia villosa*, and *Vigna* spp.

Wild Hosts:

Aeschynomene indica (Indian jointvetch), *Fabaceae* (legumes), *Geranium* (cranesbill), *Lamium amplexicaule* (henbit deadnettle), *Penstemon*, *Sesbania exaltata*, *Stellaria media* (common chickweed), and *Verbascum thapsus* (Aaron's-rod).

Known Distribution

H. glycines probably evolved either in China or Japan and from there has spread to the New World. It is now widely distributed in the U.S. (primarily the eastern half), China, and Japan, particularly in areas where soybean is grown on a commercial scale. *H. glycines* is still spreading into new areas with recent records in South America, for example. The nematode appears widespread in Brazil. Other countries where SCN is present include: Canada, Colombia, Ecuador, Egypt, India, Indonesia, Italy, Korea, Mongolia, and Puerto Rico.

Potential Distribution Within the US

Alabama, Arizona, Arkansas, Delaware, Florida, Georgia, Illinois, Indiana, Iowa, Kansas, Kentucky, Louisiana, Maryland, Michigan, Minnesota, Mississippi, Missouri, Nebraska, North Carolina, Ohio, Oklahoma, South Carolina, South Dakota, Tennessee, Texas, Virginia, and Wisconsin.

Survey

When poor plant growth is observed in a soybean field, the possibility of SCN infestation should be considered. Nematodes may be recovered from the soil or plant roots using standard nematological techniques. If you detect a problem during the growing season, plant and soil samples should be taken. The white or yellow cysts are readily visible protruding from the roots of infected plants. Check

for cysts on the healthier plants on the edge of areas where plants with severe symptoms occur. To observe females on soybean roots, dig up plants rather than pulling them. Pulling plants can leave behind a good portion of the root system where cysts occur. Gently shake loose soil from the roots and look for cysts. When soil conditions cause a large amount of soil to remain on the roots, immerse the roots in a bucket of water for several minutes. This will wash the soil from the roots without removing the cysts.

If you suspect a nematode problem, take systemic (stratified) soil samples in the fall when nematode numbers are high. Pull 20 to 30 cores 6 to 8 inches deep from each 4 to 5 acres. Samples can be taken anytime the soil is moist and in good working condition. Send soil in a plastic bag, seal tightly to keep moist, to a qualified diagnostic laboratory. Analysis of soil and/or root samples for nematodes has the advantage that it may reveal other nematode or disease problems. Include an accurate crop history (including soybean varieties planted previously). Information about fertility, herbicides, and cultural practices can also aid in diagnosis. For comparative purposes, areas showing healthy plant growth should be sampled as well as areas in decline. Keep samples out of direct sunlight to avoid overheating. Samples may also be damaged by heat if they are kept in the trunk of a car. When storing samples, avoid extremes of heat or cold. Samples should be sent to the laboratory as soon as possible after collection.

Key Diagnostics

The presence of the species would normally be confirmed by examination of the cysts once these have been extracted from the soil or removed from the roots. In practical terms, cyst nematodes recovered from fields where soybeans have been grown are assumed to belong to *H. glycines.* Molecular probes were developed by Besal et al. (1988).

H. glycines is superficially similar to other members of the genus *Heterodera*. Species differentiation within this genus can be difficult and is best left to experienced individuals. Characters used include the dimension of the cyst, structure of the vulval cone and its associated features and second stage juvenile morphometrics and morphology.

Rotylenchulus macrodoratus

Scientific Name
Rotylenchulus macrodoratus Dasgupta, Raski, and Sher

Common Name(s)
Reniform nematode

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Secernentea, **Order:** Tylenchida, **Family:** Hoplolaimidae

Reason for Inclusion in Manual

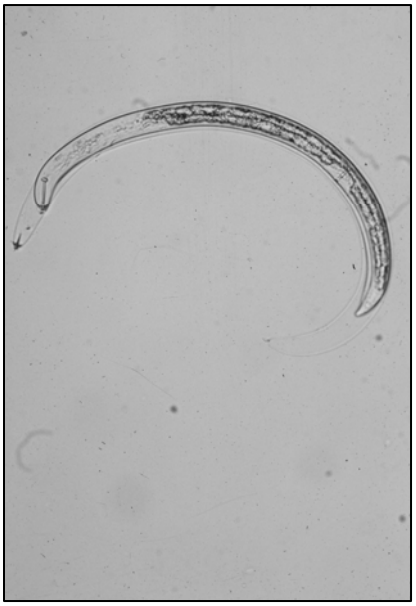


Figure 1. Vermiform *R. macrodoratus* female. Photo courtesy of Nikos Volvlas.

Pest Description
Among the species of *Rotylenchulus* of major economic importance, *Rotylenchulus reniformis* and *R. parvus* are worldwide in distribution. *R. macrodoratus* occurs only in the Mediterranean region, particularly in France, Greece, Israel, Italy, and Malta.

This reniform nematode has semi-endoparasitic sedentary habits. Single-cell *R. macrodoratus* eggs measured 111 μm (98 to 119) x 44 μm (40 to 49), about twice as long as eggs of *R. parvus* (56 to 59 μm x 30 to 38 μm). The first stage juvenile appeared after 11 to 14 days, the second stage juvenile after 14 to 17 days, and hatching occurred 16 to 19 days after egg deposition. Second stage juveniles (J2) and following juvenile stages (J3 and J4) develop and attain the adult stage in the soil without feeding (Inserra and Vovlas, 1979).

The infective stages of *R. macrodratus* were the immature females, as reported for *R. parvus* and *R. reniformis.* The vermiform females (Fig. 1) penetrate host roots and become sedentary. Immature females were found in roots 14 to 16 days after inoculation. The anterior portion of their body remains embedded in the roots and the posterior portion protrudes from the root surface and swells. They establish a specialized feeding site (a mononucleate giant cell) in the stele.

Swollen semi-endoparastic females (Fig. 2) were observed 25 to 31 days after inoculation, and 4 to 5 days thereafter fully developed females with the first eggs were found. After gonad maturation they deposit eggs in a gelatinous matrix (Fig. 2), which surrounds the female posterior body (Robinson et al., 1997). The complete lifecycle from egg to egg took about 45 to 55 days, somewhat longer than that of *R. parvus* (27 to 36 days) and more than twice that for *R. reniformis* (17 to 23 days) (Inserra and Vovlas, 1979).

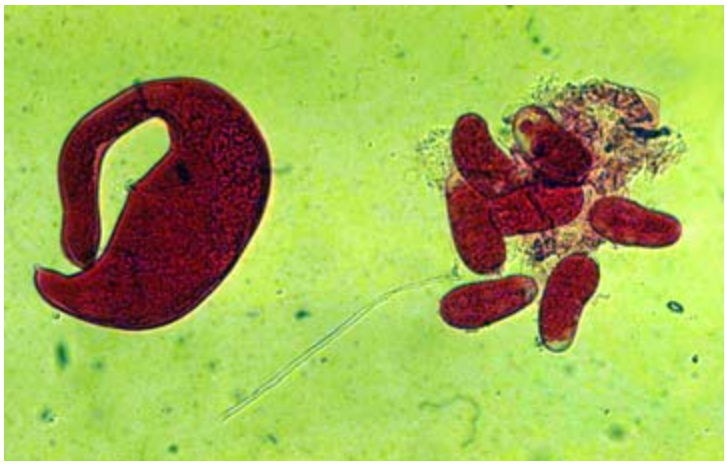


Figure 2. Adult female and eggs of *R. macrodoratus*. Photo courtesy of Nikos Volvlas.

Pest Importance

This nematode is common in the Mediterranean regions, where it parasitizes the root systems of fruit trees and various ornamentals.

Symptoms

Small swellings in the area of nematode penetration were noted in infested roots of *Dianthus* species. The symptom was not found on other hosts tested (Inserra and Vovlas, 1979). The detrimental effects of this nematode on the growth and yield of its economic hosts are unknown. Further studies are needed on the pathogenicity threshold limits and influence of population densities of this nematode on host-plant growth.

Known Hosts

This reniform nematode parasitizes many fruit crops and ornamental trees such as *Ceratonia siliqua* (carob), *Eurybotria japonica* (loquat), *Ficus carica* (fig), *Laurus nobilis* (laurel), *Nerium oleander* (oleander), *Olea europaea* (olive),*Prunus amygdalus* (along), *Pistacia vera* (pistachio), *Prunus armeniaca* (apricot), *Prunus domestica* (plum),*Vitis vifera* (grape), *Quercus calliprinos* and *Q. farnetto* (oak). Herbaceous hosts include *Dianthus barbatus (*large-flowered sweet William*), Dianthus caryophyllus* (carnation), *Glycine max* (soybean), *Hedera ile* (ivy), *Parietaria officinalis (*pellitory*),* and *Phlomis fruticosa* (phlomis).

Known Distribution

R. macrodoratus is a Mediterranean species, which occurs in France, Greece, Israel, Italy, and Malta (Robinson et al., 1997). It has recently been reported in South Africa (Van den Berg, 1998).

Potential Distribution Within the US
No information available at this time

Survey
Soil and root samples were collected for *R. macrodoratus* and other nematode pests of olive by Tedeschini et al. (2002). To collect samples, the groves were divided into sampling blocks representing differences in soil texture, drainage patterns, or cropping history. A sample of 1 to 2 kg of soil and 10 g of roots was taken for nematode analysis from 5 to 20 subsamples collected. A sample of 100 ml of soil and 10 g of root was mixed and analyzed. Nematodes from soil samples were extracted by Oostenbrink's elutriator and the root samples by centrifugation. Nematodes were killed by heat and fixed in TAF (formalin and triethanolamine). For identification, temporary and fixed mount slides were prepared.

Key Diagnostics
The morphological characteristics of the vermiform stages of this reniform nematode are similar to those of *R. reniformis*. *R. macrodoratus* vermiform females, however, have a longer stylet than those of *R. reniformis* (22 to 26 vs. 16 to 21 μm). *R. macrodoratus* swollen females lack the characteristic spike-like mucro, which is present at the body posterior end of *R. reniformis* females.

Rotylenchulus reniformis

Scientific Name
Rotylenchulus reniformis Linford and Oliveira

Synonyms:
Leiperotylenchus leiperi, Rotylenchulus leiperi, *R. queirozi*, *R. stakmani*, *Spyrotylenchulus queirozi*

Common Name(s)
Reniform nematode

Type of Pest
Nematode

Taxonomic Position
Class: Secernentea, **Order:** Tylenchida, **Family:** Hoplolaimidae

Reason for Inclusion in Manual

Pest Description
Linford and Oliviera established the genus *Rotylenchulus* in 1940 with *Rotylenchulus reniformis*, the reniform nematode, as the type species. The generic name was given by Linford and Oliviera because they thought that the nematode species was similar to the genus *Rotylenchus*. The species name was coined because of the 'kidney- shape' of the mature female (Fig. 1).

R. reniformis is a soil inhabiting semi-endoparasitic (partially inside roots) species in which females penetrate the root cortex, establish a permanent feeding site (synctia) in the stele region of the root, and become sedentary or immobile. The anterior portion (head region) of the body remains embedded

Figure 1: Mature stained female on plant root (Top), unstained females on plant roots. Photos courtesy of CABI, 2004, and H. Ferris, UC Davis.

in the root whereas the posterior portion (tail region) protrudes from the root surface and swells during maturation (Fig. 1). The reniform nematode occurs in many different non-flooded soils, but is more frequently found in heavier clay silts and clays in contrast to the root knot nematode.

Immature females: Body vermiform, slender, and spiral to C-shaped when heat killed. The length ranges from 0.34 to 0.42 mm. Stylet (16 to 18 μM) knobs are rounded and slope posteriorly. The median bulb of the esophagus has a distinct valve and the basal glands of esophagus overlap the intestine laterally and ventrally. The vulva is not prominent and occurs at about 70% of the body length. Ovaries are paired and opposed with double flexure. Tail tapers to a narrow rounded terminus.

Mature female: Body swollen, kidney-shaped, with an irregular neck. The length ranges from 0.38 to 0.52 mm. The vulva has raised lips. The body beyond the anus is hemispherical with a slender terminal portion 5 to 9 mm long. Well-developed stylet. Cuticle think. Ovaries very long, convoluted; vulva post-equatorial. Eggs deposited in a gelatinous matrix.

Figure 2: Life stages of reniform nematode, *Rotylenchulus reniformis*. From left to right is egg, juvenile, young female with swollen body, and mature female in kidney shape. Photo courtesy of Koon-Hui.

The female reproductive system is amphidelphic with two flexures in immature females and highly convoluted in mature females. The female tail is usually more than twice the anal body diameter. The juvenile tail tapers to a narrow, rounded terminus with about 20 to 24 annules. Phasmids are porelike, about the body width or less behind anus.

Male: Vermiform. The length ranges from 0.38 to 0.43 mm. Anterior end reduced; stylet reduced. The esophagus is degenerate with reduced median bulb and valve. Males do not feed. The spicules are elongate-slender, ventrally curved. Caudal alae present but difficult to see, not quite reaching tail end. Juveniles and males remain in soil.

Biology and Ecology

R. reniformis life stages are shown in Figure 2. Single-celled eggs hatch one to two weeks after being laid. *R. reniformis* has four juvenile stages, an immature female, and mature female/male stages. The first-stage juvenile molts within the egg, producing the second-stage juvenile (J2) that emerges from the egg. Further molts occur producing the third and fourth juvenile stages, all of them retaining the cuticles of the previous stages. None of these stages are parasitic,

and they do not feed on plant roots. The final molt produces an immature vermiform female or male. The vermiform immature female is the infective stage and it partly penetrates the cortex of the host plant root. A permanent feeding site in the root endodermis is developed at the head of the nematode, and it becomes sedentary. Once root penetration occurs, one or two weeks are required for females to reach maturity. The male, which remains outside of the root, can inseminate prior to the female gonad maturation, and sperm are stored in the spermatheca. Numbers of females and males in a population are usually equal. Some populations of reniform nematodes reproduce parthenogenetically (egg production without fertilization). Soon after female gonad maturation, the eggs are fertilized with sperm, and eggs are then deposited into a gelatinous matrix with about 60 to 200 eggs. The life cycle from egg to egg can be as short as three weeks, but the length of the cycle is affected by the host and environmental conditions, in particular soil temperature. It can, however, survive at least two years in the absence of a host in dry soil through anhydrobiosis (a survival mechanism without water) (Fig. 3).

Figure 3. Reniform nematode tightly coiled to undergo anhydrobiosis under drought conditions. Photo courtesy of Koon-Hui Wang, University of Florida.

Only females infect plant roots. Nurse cells form near pericycle (100-200 per female in soybean). Nurse cell system is stimulated by feeding, which causes hypertrophy of pericycle and endodermal cells, increased cytoplasm density, but cells remain uninucleate with large nucleolus. Walls may rupture to form a synctium. A syncytial cell is a multinucleated cell resulting from cell wall dissolution of several surrounding cells. Syncytium about 2 cells deep may extend half way around the root in soybeans. Syncytia are stimulated primarily in pericycle tissues (phase 1: cell wall lysis; phase 2: anabolic phase-increase in organelles in affected cells.). The nematode also feeds on cortex of cowpea and phloem of cotton.

Pest Importance

Among the crops most severely affected by reniform nematode are upland cotton, pineapple, and many vegetable crops including tomato, okra, squash, and lettuce. The university extension services in Mississippi and Alabama recommend nematicide treatment for cotton fields if population density exceeds 2 nematodes/cm^3 soil in the spring and 10 nematodes/cm^3 in the fall or winter. Besides direct damage, reniform nematodes are also an important factor in the incidence of *Fusarium* and *Verticillium* wilts of cotton, causing the Fusarium-wilt tolerant varieties of cotton to become susceptible. In Louisiana, reduction in cotton yield due to *R. reniformis* damage has been assessed as high as 40 to

60%, with a concomitant increase in Fusarium wilt, although the average is lower at 15 to 30% (Robinson et al., 1997). Reniform nematode has also reduced snap bean yield by 10% in south Florida.

In Hawaii, heavy infestations of *R. reniformis* combined with moisture stress can result in complete ratoon failure of pineapples (Caswell et al., 1990). Economic threshold for reniform nematode on pineapple is 310 nematodes/250 cm^3 of soil.

R. reniformis is a quarantine risk in all subtropical and tropical countries and also in warm temperate regions where susceptible host crops are grown. It is disseminated on the roots of host plants and in soil either in potted plants or as bare rooted seedlings (CABI, 2004). Areas free of reniform nematode impose regulations against this nematode. Chile and Switzerland are among the countries that have quarantine against reniform nematode. In the U.S., Arizona, California, and New Mexico restrict reniform nematode to protect their cotton industries. The ornamental industries of southern Florida and Hawaii are adversely affected by this regulation when the plant shipments are contaminated with reniform nematode. Therefore, expensive sanitation practices and the use of clean materials are required for ornamental plant nurseries (Wang, 2001).

Figure 4: Severe stunting of cotton by the reniform nematode. Photo courtesy of LSU Ag Center.

Symptoms/Signs

Reniform nematode damage is often difficult to diagnose in the field. Infected plants exhibit various degrees of stunting (Fig. 4), signs of nutrient deficiency (leaf chlorosis) associated with an impaired root system, reduced yield, and often early maturity. Above ground symptoms on the host plant also include shedding of leaves and formation of malformed fruit and seeds. Below ground, roots can range from generally healthy, to being smaller and sparse, to being discolored and necrotic with areas of decay. Plant mortality is possible in heavy infestations.

The most economically important pathogen in the pineapple industry of Hawaii and the Philippines is the reniform nematode *R. reniformis* (Starr and Page, 1990). Infected pineapple plants show poor growth, reddish leaves, and are less upright than healthy plants.

Seedling emergence is delayed in some vegetables, tobacco, and legumes. In sweet potato, reniform nematodes may cause surface cracking of tubers. In cotton, symptoms resemble those caused by root knot nematode. Patchy growth typical of root damage is common, and stunting, leaf chlorosis, and wilting can also occur. Reniform nematode does not cause galling of roots but causes reduction in root growth. In young tea plants, *R. reniformis* feeds mostly on the fine feeder; above-ground symptoms of infestation are similar to those caused by other nematode species (CABI, 2004).

Known Hosts

R. reniformis has an extremely wide host range covering most of the plant families. The reniform nematode attacks over 140 species of more than 115 genera in 46 families. However, most of the known hosts are secondary, and pest damage is either minor or has not been investigated. The nematode is recognized as an economically important damaging pest particularly on cotton, pineapple, sweet potato, and soybean. There are conflicting reports on some plants that have been described both as hosts and non-hosts including: *Allium* spp., *Brassica* spp., *Citrus*, coffee, and rice. The existence of biological races of the reniform nematode may partly explain these observed differences in host range.

Major hosts

Abelmoschus esculentus (okra), *Ananas comosus* (pineapple), *Brassica oleracea* var. *capitata* (cabbage), *Cajanus cajan* (pigeon pea), *Citrus* spp., *Cucumis melo* (melon), cucurbits, *Glycine max* (soybean), *Gossypium* (cotton), *Ipomoea batatas* (sweet potato), *Lycopersicon esculentum* (tomato), *Musa* (banana), *Phaseolus* (beans), *Solanum melongena* (eggplant), *Syzygium aromaticum* (clove), and *Vigna ungiculata* (cowpea).

Known Distribution

R. reniformis is largely distributed in tropical, subtropical, and in warm temperate zones. Countries where the nematode is present include: **Asia:** Bangladesh, China, India, Indonesia, Iraq, Israel, Japan, Lebanon, Malaysia, Oman, Pakistan, Philippines, Singapore, Sri Lanka, Thailand, Vietnam; **Europe:** Greece, Malta, Spain; **Africa:** Angola, Burundi, Cameroon, Cape Verde, Egypt, Gambia, Ghana, Kenya, Liberia, Malawi, Mozambique, Nigeria, Reunion, Senegal, Somalia, South Africa, Sudan, Tanzania, Togo, Uganda, Zimbabwe; **North America:** Mexico, United States; **Central America:** Antigua and Barbuda, Barbados, Belize, Bermuda, Costa Rica, Cuba, Dominica, Dominican Republic, Grenada, Guadaloupe, Honduras, Jamaica, Martinique, Montserrat, Panama, Puerto Rico, Saint Lucia, Saint Vincent and the Grenadines, Trinidad and Tobago; **South**

America: Brazil, Colombia, Guyana, Peru, Suriname, Venezuela; **Oceania:** American Samoa, Australia, Fiji, Guam, Kiribati, Niue, Papua New Guinea, Samoa, Solomon Islands, and Tongo.

Potential Distribution Within the US

R. reniformis was first found on cowpea roots in Hawaii (Linford and Oliveira, 1940) and first reported as a parasite of cotton in Georgia and of tomato in Florida. Today, it is found throughout the southern U.S. Within the U.S., the reniform nematode is known to be established in Alabama, Arkansas, Florida, Georgia, Hawaii, Lousiana, Mississippi, Missouri, North Carolina, South Carolina, and Texas. Its reported pattern of distribution suggests that it is likely to be present in southwestern Tennessee and possibly Oklahoma. In California, *R. reniformis* infected *Phoenix roeselenii* and *Cycas* spp. plants were detected in San Diego in 1960, having entered the state in a quarantine shipment. The plants had been established in a residential property before a confirmed diagnosis of the pest had been completed. Subsequently, the plants were removed from the infested site and fumigated with methyl bromide. The planting site was also fumigated with methyl bromide (Ferris,1999).

Survey

R. reniformis is a microscopic organism found in soils and roots. When roots are severely infested with the nematode, they can appear dirty because of soil particles adhering to the gelatinous matrices of the nematodes on the surface of the root. To accurately determine its presence or association with disease symptoms, the nematode has to be extracted from the soil or roots by standard nematode extraction procedures. Identity of the extracted nematodes is confirmed by microscopic examination. All stages of the nematode up to the immature female and mature male can be extracted from soils. Root extractions will mainly provide hatched second stage juveniles from the eggs on the root surface and possibly some immature females.

Gazaway and McLean (2003) collected composite soil samples from 8 hectares in each field. Nematodes (*R. reniformis*, *Meloidogyne incognita*, and *Hoplolaimus columbus)*, were extracted by gravity screening and sucrose centrifugation, identified to genus using microscopy, and quantified. Each composite sample consisted of 20 soil cores (2.5-cm-diam. X 20-cm deep) taken in a systematic, zig-zag sampling pattern from an 8-ha section of each field. Composite samples were sealed in plastic bags and stored in a cool ice chest (less than four hours) as they were collected from the field until being transferred to a 5°C refrigerator prior to extraction. Each sample was thoroughly mixed and a 100 cm^3 sub-sample was collected for nematode extraction,

Key Diagnostics

There are ten species in the genus *Rotylenchulus*. *R. reniformis* is the most economically important and widely distributed species and, therefore, the one most likely to be encountered. It is similar in general respects to the other

members of the genus, but most easily differentiated by the shape of the obese mature female, particularly the characteristic hemispherical posterior body with a terminal spike. The mature female is also similar in superficial respects to *Achlysiella*, but can be distinguished by the more posterior vulva, the differently shaped lip region, the more ventrally overlapping oesophageal gland lobes and the form of the posterior body.

Xiphinema ifacolum

Scientific Name
Xiphenema ifacolum Luc

Common Name(s)
Dagger nematode

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Adeenophorea, **Order:** Dorylaimida, **Family:** Longidoridae

Reason for Inclusion in Manual

Pest Description
Species in the genus *Xiphinema* are relatively large nematodes, 2 to 3 mm in length (Fig. 1). The genus is characterized by the presence of a very long odontostyle or spear (stylet). The spear and its extension are approximately 150 µm or more in length.

Xiphinema ifacolum was first described by Luc in 1961. The type population came from soil about the roots of *Citrus vulgaris* (*C. aurantium*) growing at the IFAC research station in French Guinea. Since that time, it has remained a valid species and is distinctive because of the tail shape and internal structure and the presence of a Z-organ in each genital tract. Even so, the assistance of a taxonomist is recommended to confirm the identification.

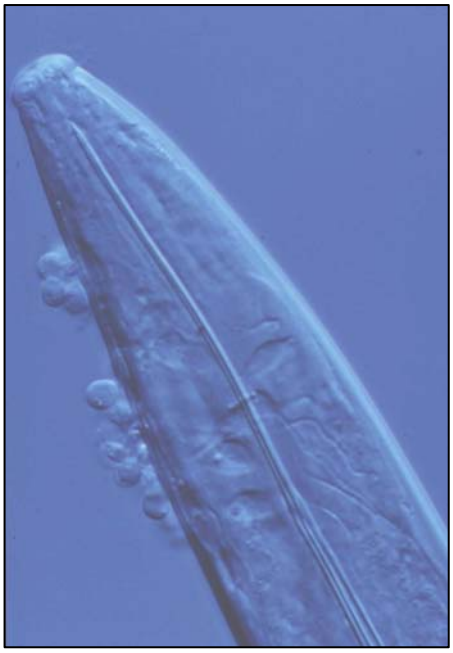


Figure 1. *Xiphinema* spp. Photo courtesy of H. Ferris, University of California, Davis.

Female: Habitus open 'C'-shaped. Body slender, cylindrical; tapering at either end. Cuticle very finely striated transversely. Lateral chord occupying one quarter of corresponding body diameter near middle of body. Latero-subventral, latero-subdorsal and cervical body pores present. Ventral pores present in posterior region of body. Head region rounded, separated from body by a weak depression. Amphidial aperture opening at level of depression and occupying about two-thirds of corresponding body diameter. Odontostylet 185 to 197 µm long; odontophore 52 to 79 µm long. Oesophagus typical of the genus. Vulva median. Genital tracts amphidelphic with very prominent Z-organs. Tail irregularly conoid, narrowed towards the tip with a subdigitate process. Three pairs of caudal pores. A narrow, characteristically shaped protoplasmic process extends down the subdigitate portion of the tail. L=3.12 to 3.71 (3.49) mm; a=50.3 to 62.0; b=7.3 to 11.3; c=45.4 to 59.1; V=(12.2 to 31.6) 48.3 to 53.3 % (12.5 to 23.3) (Luc, 1961).

Male: Similar to the female in general body form. Spicules massive, broad and arcuate; measuring 53 µm along the chord. Supplements arranged as one ventral pair just before the cloaca and three others ventral plus five subventral pairs. Tail similar to the female in shape with three pairs of caudal pores. Male (allotype): L=3.20 mm a=62.9; b=6.7; c=43.9; T=? (Luc. 1961).

Biology and Ecology

X. ifacolum is a migratory ectoparasite of plant roots. As with other longidorids, it is polyphagous. There are four juvenile stages plus the egg and adult. Coiro et al. (1995) discussed the fecundity and longevity of *X. ifacolum* on tomato. They found that under laboratory conditions, females had a longevity of about 34 weeks at 25°C and produced 82 to 90 progeny over a 20-week span. No other information on the biology and ecology of *X. ifacolum* is available at this time.

Pest Importance

Several reports suggest that *X. ifacolum* is capable of causing damage to rice in Liberia (Lamberti et al., 1987, 1991) and to black pepper in Sri Lanka (Lamberti et al., 1983). Lamberti et al. (1987) suggested that synergistic interactions with other pathogens could suppress growth of rice in Liberia, and subsequently regarded the nematode as a major pest of rice in this country. Pot experiments with rice indicated a 30% growth reduction in the presence of *X. ifacolum* (Lamberti et al., 1987). Lamberti et al. (1993) reported *X. ifacolum* as pathogenic to soybean. Lamberti et al. (1992b) reported reproduction of *X. ifacolum* on, and pathogenicity to, cowpea, okra, pepper and eggplant in Liberia. Assessments of economic loss are not available.

Symptoms/Signs

Ectoparasitic feeding on roots causes root tip swelling and galling with localized discoloration and necrosis. Root growth is suppressed and root systems reduced in size. It may be associated with declining plants and patchy growth of rice in the field with lower yield of grain (Lamberti et al., 1987, 1991). As with most plant

parasitic nematodes, the effect on the above ground parts is indirect and due to destruction of the root system. The species is not known to transmit viruses.

Known Hosts

X. ifacolum has been recovered from the rhizosphere of a wide variety of plants and is particularly associated with rice. It is known to attack the roots of tomato in pot experiments. Other reports indicate soybean, cowpea, eggplant, okra, pepper, cocoa, citrus and coffee as hosts. As with other members of the genus, this species is likely to be polyphagous.

Major hosts

Oryza sativa (rice)

Minor hosts

Abelmoschus esculentus (okra), *Ananas comosus* (pineapple), *Capsicum annuum* (bell pepper), *Citrus limon* (lemon), *Coffea* (coffee), *Glycine max* (soybean), *Hevea brasiliensis* (rubber), *Lycopersicon esculentum* (tomato), *Musa* (banana), *Musa x paradisiaca* (plantain), *Piper nigrum* (black pepper), *Solanum melongena* (eggplant), *Theobroma cacao* (cocoa), and *Vigna unguiculata* (cowpea)

Known Distribution

X. ifacolum is widely distributed in West Africa and has also been recorded from South America (Brazil) and Sri Lanka. It is likely that this species was originally an inhabitant of virgin equatorial rain forest, at least in West Africa and South America. The species has also been recorded from Benin, Brazil (Para State), Cameroon, Guinea, the Ivory Coast, Togo and Sri Lanka.

Potential Distribution Within the US

No information is known at this time.

Survey

X. ifacolum may be recovered from soil using standard extraction techniques, but those involving sieving (particularly immersion sieving) will be more efficient, particularly for recovery of the adult stages.

Key Diagnostics

X. ifacolum is broadly similar to other members of the genus *Xiphinema*, an extensive assemblage of over 250 nominal species. It is readily distinguished by the presence of Z-organs in the female genital tracts and by the form of the tail and the characteristic protoplasmic extension down the subdigitate portion.

Parasitic Plants/Weeds

Alectra vogelii

Scientific Name
Alectra vogelii Benth.

Synonyms:
Alextra angustifolia, Alextra merkeri, Alextra scharensis

Common Name(s)
Alectra, yellow witchweed, cowpea witchweed

Type of Pest
Hemiparasitic plant

Taxonomic Position
Class: Magnoliopsida, **Order:** Scrophulariales. **Family:** Scrophulariaceae

Reason for Inclusion in Manual

Pest Description
Flowers: Flowers are five-lobed, sulfur yellow to pale orange (Fig. 1), bell shaped with large horseshoe shaped stigma. Plant height ranges from 30 to 45 cm tall, often as a single stem, but sometimes branch near ground level. Flowers are borne individually on short stems in the axils of the upper leaves. The corolla, formed of five petals which are fused into a tube for the bottom half, is bell-shaped when open, 1.6 to 1 cm in diameter, and somewhat longer than the calyx. Petals are generally pale yellow and may or may not have three deep red veins. Both flower forms can be found in the same stand of *A. vogelii*. Anthers and filaments are glabrous. After flowering, the corolla withers and remains covering the developing globose seed capsule, which eventually swells to approximately 5 mm in diameter.

Leaves: Leaves are 1.5 to 3.5 cm long by 0.3 to 1.5 cm wide and are hairy. Leaf margins vary from five or six sharp teeth to two to five widely spaced teeth, with some plants having entire margins.

The chromosome number (2n) is 38.

Pest Importance

Alectra vogelii is a parasitic weed found in major leguminous crops, including chickpea, cowpea, soybean, and runner bean. In 1929, one report estimated a 20% loss in yield for cowpea crops in Kenya. In 1966, the Agricultural Department for Botswana reported a loss of 24,000 acres in cowpea due to 'yellow witchweed'. In 1977, on-farm trials in Botswana produced no cowpea yields in 6 out of 25 blackyeye crops. In 1979, a blackeye cowpea trial had an average yield of 602 kg/ha and 100 kg/ha for the non-infested and infested fields, respectively. Yield losses of 15% are reported for peanut production in Nigeria, and a 30 to 50% reduction in bambara nut yields in South Africa. A ten year crop rotation study found that long-term rotation with non-crop hosts did not reduce the density of *Alectra* infestations.

Figure 1. Mature *A. vogelii* plant (left) and close up of flowers (right). Photos courtesy of C.R. Riches (CABI, 2004).

Symptoms/Signs

Symptoms associated with *A. vogelii* include: stunted crop plants with smaller leaf area, shorter leaf petioles, and increased shoot/root ratios. Roots are bright orange below soil surface. Stems and leaves are conspicuously hairy. The dust-like seeds have a complex structure. An outer cell layer of the testa is modified into a cone or a 'trumpet-like' structure about 1 mm long, within which the 'kernel' of the seed, measuring about 0.15 mm by 0.25 mm, is suspended. The surface of the seed coat is covered in indentations.

Known Hosts

Major hosts

Vigna unguiculata (cowpea)

Minor hosts

Arachis hypogaea (peanut), *Glycine max* (soybean), *Lablab purpureus* (hyacinth bean), *Mucuna pruriens* (Buffalobean), *Phaseolus acutifolius* (tepary bean), *Phaseolus coccineus* (runner bean), *Phaseolus radiata*, *Phaseolus vulgaris* (common bean), and *Voandzeia subterranea* (bambara groundnut)

Wild hosts

Acanthospermum hispidum (bristly starbur)

Known Distribution

A. vogelii is distributed throughout semi-arid areas of tropical Africa and subtropical southern Africa, from Swaziland and South Africa in the south, to Burkina Faso and Mali in the west, to Kenya in the east. This species is closely associated with cropping and is rarely found in natural areas. *A. vogelii* is distributed throughout semi-arid areas of tropical and sub-tropical Africa. In the Nigerian savannahs, it can be found in cowpea crops, which are also attacked by *Striga gesnerioides*, and it has been reported as the major parasite of the crop in the northern Guinea savannah (Lagoke, 1989). Elsewhere in West Africa, infestations tend to be more localized, as in southern Mali. *A. vogelii* has replaced *S. gesnerioides* as an important constraint to cowpea production in eastern, central, and particularly southern Africa.

Potential Distribution Within the US

Information is not available at this time.

Survey

Plant type: Annual; vine/climber; shrub; herbaceous; seed propagated. Flowers are five-lobed, sulfur yellow to pale yellow, and bell-shaped. Hairy stems and leaves on parasitic weed, combined with stunted crop plants.

As *A. vogelii* is largely dependent on annual cropping, environmental requirements mirror those of its major hosts cowpea, bambara, peanut and soybean in sub-Saharan Africa. By and large, infestations are found in semi-arid areas with a short growing season of 4 to 6 months, below 1500 m altitude. The parasite is most commonly found in areas of mono-modal rainfall with a long dry season as in Botswana or the Guinea savannah of West Africa, but it is also a pest in bimodal rainfall areas as in north-west and coastal Tanzania. Although crops are not produced during the cold dry season in the range of the parasite, frost at the end of the growing season will kill host plants surviving in crop residue on residual moisture and will prevent further seed production by *A. vogelii.* Host crops are largely associated with free-draining sands and sandy-loams.

Climatic amplitude (estimates):
- Mean annual rainfall: 520 to 1000 mm
- Rainfall regime: summer; bimodal
- Dry season duration: 6 to 7 months
- Mean annual temperature: 19 to 26ºC
- Mean maximum temperature of hottest month: 29 to 38ºC
- Mean minimum temperature of coldest month: 6 to16ºC
- Absolute minimum temperature: -3 to 0ºC

Borreria latifolia

Scientific Name
Borreria latifolia

Synonyms:
Borreria alata, Borreria bartilingiana, Borreria perrottettii, Borreria scaberrima Spermacoce coerulescens, Spermacoce alata, Spermacoce latifolia

Common Name(s)
Broadleaf buttonweed

Type of Pest
Weed

Taxonomic Position
Class: Dicotyledonae, **Order:** Gentianales, **Family:** Rubiaceae

Reason for inclusion in manual

Pest Description
Borreria latifolia (Fig. 1) is a branched herb, prostrate, ascendent or erect, usually branched from the base, stems fleshy, 4-winged, about 75 cm tall; leaves opposite, elliptical, broadest above the middle, tip broadly and shortly pointed, base tapered, variable in size about 2.5 to 5.0 cm long and 2.5 cm wide, thick, hairy on both sides, short leafstalk; leaf base joined with cup-shaped stipules with bristles on edges. Inflorescence in leaf axils, 0.6 to 1.2 cm across, off white, each flower with hairy calyx of four sepals; stamens 4 and stigma forked; flowers throughout the year; fruit hairy, splitting into two pairs to release seeds.

Hypocotyl 15 to 23 mm long, papillate, reddish green. Cotyledons 2; stipules hairy, inter-petiolar; petiole 2 to 2.5 mm, glabrous, green to reddish green, blade broadly ovate, 9.5 to 11.5 by 9.5 to 10 mm, glabrous, mid-nerve distinct, base obtuse, margin entire, apex shallowly emarginate. Epicotyl 3 to 4.5 mm long, 4-winged and hairy. First leaves 2, with inter-petiolar stipules; stipule 3-lobate, hairy; petiole about 1 mm long, densely hairy; blade ovate to narrowly ovate, 10.5 to 19.0 by 5 to 8.5 mm, short and hairy, pinnately nerved, based attenuate to obtuse, margin entire, short and hairy, apex subacuminate.

Biology and Ecology

B. latifolia grows well in humid tropical regions with a short and pronounced dry season, on sunny or lightly shaded shallow fields or those with a second crop, along roads and steep riverbanks (Soerjani et al., 1987). It also grows on poor soils and prefers sandy soils (Soerjani et al., 1987). It is found up to an elevation of 1600 meters in Thailand (Harada et al., 1987).

B. latifolia reproduces by seed. It is a prolific seeder, but there has been no determination of its reproductive capacity. It requires light to germinate. Once established, it is fast growing and becomes reproductive within 2 months. The weed is palatable to domestic animals, such as cattle, goats and chickens. However, there is no information on the survival of the seeds after they pass through the alimentary canal of these animals.

Seeds that have been extracted from soils from oil palm and rubber plantations were 26 to 29% viable. In rubber, the seed density was 26 to 70 per m^2 but was 242 to 553 per m^2 in oil palm. It favors more open conditions. This is evident when flushes of seedlings emerge after the destruction of the previous vegetation by glyphosate and glyphosate mixtures. *B. latifolia* can also spread vegetatively.

Figure 1. Stem and foliage (left) and growth pattern of *B. latifolia* (right). Photos courtesy of Chris Parker and Novartis (CABI, 2004).

Pest Importance

B. latifolia at high densities competes with crops for nutrients and water. It reduced the dry weight and height of young rubber by 12 and 17%, respectively. Together with other species, the critical period of competition in rubber is 4 to 6 weeks after transplanting. In upland rice, the critical period of competition is 4 to 8 weeks after sowing. However, it is not known to affect the growth of tea.

Symptoms/Signs

B. latifolia competes with crops for nutrients and resources and has the potential to reduce crop yield.

Known Hosts

Major hosts
B. latifolia is a common weed in sugarcane, rubber, oil palm, orchards, tea, chinchona, cassava and many annual upland crops such as maize, soybean and rice. Other hosts include *Allium cepa* (onion), *Gossypium* spp. (cotton), *Theobroma cacao* (cocoa), and *Vigna radiata* (mung bean).

Known Distribution
B. latifolia originated from the West Indies and tropical America, but now has a pan-tropical distribution in 19 countries. *B. latifolia* is a common weed in Indonesia, Malaysia and Thailand. Introduced in Java, it has become naturalized in Sumatra, Java, Kalimantan and Sulawesi. In Malaysia, it is distributed throughout the Peninsula and is found in Sarawak and Sabah. It is also widely distributed in Thailand. Other countries with the weed present include: Bhutan, Brunei Darussalam, Cambodia, India, Nepal, Singapore, Sri Lanka, Vietnam, Ivory Coast, Ghana, Guinea, Liberia, Nigeria, Senegal, Sierra Leone, Uganda, Mexico, Costa Rica, Netherlands Antilles, Panama, Bolivia, Brazil, Suriname, Australia, and French Polynesia.

Potential Distribution Within the US
Information is not available at this time.

Survey
Annual herb. Stems: prostate 20 to 40 cm, square 4-winged, hairy. Leaves: simple, opposite, shortly petioled, broad elliptic, acute apex, penninerved, adaxial scabrous. Flowers: clusters, axillary, light pink/rarely 4 white lobes. Fruits: capsule, subglobose, wrinkled, hairy, split longitudinally. Seeds: ellipsoid, brownish, 2 to 3 mm long. Reproduces by seeds.

Key Diagnostics:
B. latifolia may occur together with *B. laevis*. However, the two weeds can be distinguished on stem morphology. The former has stems conspicuously winged and the latter has stems 4-ribbed or 4-angled but not distinctly winged (Soerjani et al., 1987). Further, the stem of *B. latifolia* is succulent while that of *B. laevis* is wiry. Salamero et al. (1997) published a key to weeds of West Africa, which can help distinguish *B. latifolia* from several somewhat similar species.

Salamero et al. (1997) provide a valuable guide to 13 related weeds in the Rubiaceae occurring in West Africa, based on vegetative characters. The two species closest to *B. latifolia* vegetatively are *Spermacoce ocymoides* (syn. *Borreria ocymoides*), which differs in generally having smaller, more slender stem and leaves with stem angle scabrid, and white to pinkish flowers; and *Diodia sarmentosa* with only stem angles pubescent, instead of hairy all round as in *B. latifolia.*

Commelina benghalensis

Scientific Name
Commelina benghalensis Linnaeus

Synonyms:
Commelina prostrata

Common Name(s)
Wandering jew, Benghal dayflower, tropical spiderwort

Type of Pest
Weed

Taxonomic Position
Class: Monocotyledonae, **Order:** Commelinales, **Family:** Commelinaceae

Reason for Inclusion in manual

Pest Description
Commelina benghalensis belongs to a family with 500 to 600 species with distinct characteristics. *C. benghalensis* is a monocot with creeping stems (Fig. 1) which assume an ascending position, are 15 to 40 cm long, branched and rooting at the nodes. The leaves are ovate or elliptical, acuminate, 3 to 7 cm long, 1 to 2.5 cm wide with a base narrowed into a petiole. The flowers are subtended by bracts with their edges fused to a length of about 10 mm to form a flattened funnel-shaped spathe, 1.5 cm long and wide. Flowers have three lilac blue petals 3 to 4 mm long (Fig. 2), the lower

Figure 1. Aboveground foliage of *C. benghalensis*. Photo courtesy of C.R. Ramsey, USDA-APHIS.

rather smaller than the two laterals and occasionally white. There are two anterior cells, which are two-ovuled. The fruit consists of a pear-shaped capsule with five seeds and the capsule opens when mature (dehiscent). Seeds, which sometimes appear sugar-coated, are 2 mm long, ribbed-rough (rugose) and grayish brown in color. Seeds have very large and deep angled reticulations, except on one side, which bears a long, black ridge. Seed surface finely granular.

Figure 2. *C. benghalensis* flowers and leaves. Photo courtesy of S.D. Sawant (CABI, 2004).

C. benghalensis produces white underground rhizomes with reduced leaves and closed modified flowers, which produce subterranean seeds (Fig. 3). These seeds are fewer but remain viable longer than the aerial ones.

Figure 3. Belowground roots and subterranean flowers. Photo courtesy of S.D. Sawant (CABI, 2004).

Biology and Ecology

C. benghalensis is a fleshy, herbaceous, creeping annual, which becomes perennial depending on moisture conditions. It is found in wet and dry lands making it a troublesome weed in arable and plantation crops. *C. benghalensis* grows best in moist and highly-fertile soils. Stems have a high moisture content, and once well rooted the plant can survive for long periods without moisture availability (Wilson, 1981) and can then grow rapidly on the onset of rains (Holm et al., 1977). It reproduces both vegetatively and by seeds. It spreads by runners, which root at the nodes and by re-establishment of stem fragments. It also produces underground stolons, which bear cleistogamous flowers and seeds, in addition to the normal aerial flowers (Budd et al., 1979).

One *C. benghalensis* plant can produce about 1600 seeds (Pancho, 1964). Freshly shed aerial seeds have a dormancy period that depends on an impermeable seedcoat but will germinate following scarification or pricking of the seed. Aerial seeds germinate mainly from the upper 5 cm, while the larger subterranean seeds may emerge from depths down to 14 cm (Budd et al., 1979). These authors found that a majority of seedlings in the field in Zimbabwe derived from subterranean seeds. However, Walker and Evenson (1985a, b) concluded

that the aerial seeds were the more important in Queensland, Australia. They also distinguished large and small classes of seed within the aerial and subterranean, and showed each of the four classes to have characteristic germination behavior. Subterranean seeds had a more pronounced light requirement for germination and a higher optimum germination temperature (28 versus 24°C). They comment on the long persistence of the seeds due to dormancy and the corresponding difficulty of control. Fertilizer application reduced seed production and resulted in stunted growth when grown under artificial dense competition in cereals in Russia (Shcherbakova, 1974).

The rate of stem elongation, branch and leaf formation increases as the node number on the stem increases (Chivinge and Kawisi, 1989). Broken stems may persist on the soil surface for several weeks or months in low moisture conditions and easily form leaves 10 to 14 days after moisture becomes available. Though stem cuttings on the surface regenerate easily (Chivinge and Kawisi, 1989), cuttings buried deeper than 2 cm fail to regenerate (Budd et al., 1979).

Pest Importance

C. benghalensis is primarily an agricultural problem in genetically modified organism (GMO) row crops, such as Roundup Ready Cotton and Roundup Ready Soybean (Fig. 4). It is partially tolerant to glyphosate (< 55% control) and has the potential to germinate in mid to late growing season. Thus, it receives minimal control with the current Roundup Ready herbicide programs, and is rapidly becoming the dominant weed in cotton in southeast Georgia.

Figure 4. Infestation of *C. benghalensis* in a Georgia cotton field. Photo courtesy of Stanley Culpepper. 1714Hwww.invasive.org

C. benghalensis is a major host for cucumber mosaic virus (CMV). Tobacco plants can be infected with this virus, which is vectored primarily by green and red aphids. Infected tobacco plants produce wilt symptoms. The virus can be readily transmitted by the aphids into tobacco. It is reported that as *C. benghalensis* coverage increases so does the incidence of CMV. *Commelina* is an alternative host of the root-knot nematode *Meloidogyne incognita* (Valdez, 1968), of the reniform nematode *Rotylenchulus* spp. (Edmunds, 1971), groundnut rosette virus (groundnut rosette assistor luteovirus) (Valdez, 1968), and of groundnut mosaic virus (groundnut rosette umbravirus) (Adams, 1967). In the Dharwar district of India, the weed is a host of *Cuscuta*

chinensis (Awatigeri et al., 1975) and an alternative host of *Corticium sasakii*, a leaf blight of rice (Roy, 1973).

The economic importance of *C. benghalensis* is related to its persistence in cultivated lands and the difficulty associated with its control. *C. benghalensis* seriously competes with arable and plantation crops in most of Africa. It is one of the troublesome weeds that effects several crops in Eastern and Southern Africa, sugarcane in the Philippines, maize in India, Indonesia, the Philippines and Taiwan and pineapples in Taiwan and Swaziland. The affect on crop growth and yield vary with each crop and with environmental conditions. Peanut flower production may be delayed by 1 to 2 weeks and nodules are also reduced depending on the intensity of infestation. Removal of *C. benghalensis* in India increased peanut yield by 27%. The value of rice was reduced in Texas when the *C. benghalensis* seed contamination was 20 seed/kg rice.

The plant is used for medicinal purposes by many African tribes for treating sore throats, eyes and burns. In India and the Philippines, the weed is used for food during famine periods.

Symptoms/Signs

C. benghalensis competes with crop species and can have a detrimental effect on crop growth and yield.

Known Hosts

Major hosts

Agave sisalana (sisal hemp), *Ananas comosus* (pineapple), *Arachis hypogaea* (peanut), *Brassica napus* var. *napus* (rape), *Camellia sinensis* (tea), *Capsicum frutescens* (chilli), *Citrus limon* (lemon), *Citrus sinensis* (navel orange), *Coffea arabica* (arabica coffee), *Corchorus olitorius* (jute), *Glycine max* (soybean), *Gossypium hirsutum* (Bourbon cotton), *Guizotia abyssinica* (niger), *Ipomoea batatas* (sweet potato), *Lycopersicon esculentu*m (tomato), *Manihot esculenta* (cassava), *Momordica charantia* (bitter gourd), *Musa* (banana), *Oryza sativa* (rice), *Phaseolus vulgaris* (common bean), *Prunus armeniaca* (apricot), *Prunus persica* (peach), *Saccharum officinarum* (sugarcane), *Sorghum bicolor* (sorghum), *Vigna radiata* (mung bean), *Vigna unguiculata* (cowpea), *Vitis vinifera* (grapevine), and *Zea mays* (maize)

Known Distribution

C. benghalensis is a weed of the tropics and subtropics. It is widely distributed in West Africa, East Africa, Central, Southern and South-East Asia extending as far as Japan, the Philippines and Australia. *C. benghalensis* is reported as a principal weed in upland rice in India and the Philippines, tea in India, coffee in Tanzania and Kenya, soybeans in the Philippines, and cotton and maize in Kenya (Holm et al., 1977). It is also a common weed in rice in Sri Lanka, sugarcane in India, the Philippines and Mozambique; cassava in Taiwan; maize in Zimbabwe (Chivinge, 1983), Angola, India, the Philippines and Taiwan;

peanuts in Zimbabwe, India and the Philippines; pineapples in Taiwan and Swaziland; cowpeas and sorghum in the Philippines; tea and citrus in Mozambique and roselles in Indonesia; cotton in Zimbabwe (Chivinge, 1988). It is also a weed of barley, jute, sisal, beans, pastures, sweet potatoes, vineyards and cereals in many countries.

Potential Distribution Within the US

C. benghalensis is presently found in 15 counties in southern Georgia, Florida, Alabama, Louisiana, California, Hawaii, and recently North Carolina.

Survey

C. benghalensis is an annual or perennial herb with fleshy creeping stems that root readily at the nodes. It is equally abundant on all soil types and pH; grows in a wide range of habitats, varying from water-saturated to dry soils; grows rapidly and forms dense mats at the nodes under optimum conditions. *C. benghalensis* is found in arable and plantation crops, and non-crop lands. Ovate leaves are parallel veined with entire margins, with a ‘monocot’ appearance. Stems have long red and white hairs at top of the leaf sheath. Unsupported plants will have a ‘vine-like’ matted, appearance and stems/runners root at nodes. Subterranean flowers have long, white, enlarged “stems” that readily contrast with the root system.

Key Diagnostics

The species is distinguished from others by the blue flowers, the short flower stalk which does not extend above the spathe, the partially joined spathe margins and the reddish brown hairs on the leaf sheath.

Euphorbia heterophylla

Scientific Name
Euphorbia heterophylla

Synonyms:
Euphorbia geniculata, Euphorbia prunifolia, Euphorbia taiwaniana, Euphorbia zonosperma, Poinsettia geniculata, Poinsettia heterophylla

Common Name(s)
Wild poinsettia, red milkweed, Mexican fireplant, painted spurge

Type of Pest
Weed

Taxonomic Position
Class: Diocotyledonae, **Order:** Euphorbiales, **Family:** Euphorbiaceae

Reason for Inclusion in Manual

Pest Description
Euphorbia heterophylla (Fig. 1) is herbaceous, erect and 20 to 200 cm in height (depending on growing conditions). The most common size is 40 to 60 cm tall. A milky latex is present when most parts of the plant are broken. The stem is branched and cylindrical, with nodes at regular intervals. The surface is smooth and reddish-green.

Obovate to lanceolate leaves are formed along the stem, with secondary branches sprouting from axillary buds. Basal leaves are long-petiolate and alternate. Upper leaves are sessile and opposite or verticillate, forming a cluster of bracts, often with a pale patch at the base, subtending the terminal inflorescence. The latter consists of a dense cluster of small, short-stalked cyathia. Each cyathium comprises a cup-shaped involucre with inconspicuous male flowers producing a single stamen only, and a female flower, without sepals or petals, producing a 3-lobed, yellowish-green fruit.

E. heterophylla shows variation in morphological features, mainly in leaf shape.

Such variability has led to divergent opinions about the different species of the genus. Oliveira and Sa-Haiad (1988) have made taxonomic studies of *E. heterophylla* and *E. cyanthophora*. Their studies of the leaf anatomy revealed a wide variation in leaf shape, which was not related to geographical distribution. However, both species were taxonomically distinguishable, using other characteristics.

Seeds are 2.5 to 3 mm wide and 2.5 mm long, oblong to oboval and dark brown to black. The surface is pitted with transverse ridges.

The seedlings have elliptical-short smooth petiolated cotyledonous leaves, green or reddish-green. First true leaves are opposite obovate to lanceolate with an acute apex and are shiny green

Figure 1. Flowers and leaves (left) and foliage and branches of *E. heterophylla*. Photos courtesy of Novartis and Kurt Kissman (CABI, 2004).

Biology and Ecology

E. heterophylla has an annual, spring-summer cycle.

Seeds are produced in great quantities with high viability. Seeds recently shed are dormant. Light and alternate temperatures (25/3°C) stimulate germination (Kissman and Groth, 1993). Each fruit bears three seeds, which are expelled when the fruit is ripe. In Brazil, seeds germinate and seedlings emerge throughout most of the year. Seed longevity is high, and seeds may remain viable with a low dormancy level after being eaten by birds (Kissman and Groth, 1993).

E. heterophylla seeds germinate over a wide range of conditions, which explains why the plant is becoming an increasingly serious problem; germination was at least 95% when exposed to a solution of pH 2.5 to 10 or a solution with osmotic potential of up to -0.8 MPa. Light was not required for germination. Seed germination occured at temperatures ranging from 20 to 40°C with maximum germination (97%) at 35°C (Brecke, 1995). Etejere amd Okoko (1989) reported 95% seed viability. Seed weight increased by 63% after 36 hours of water

imbibition. Optimum germination of the seeds occurred at 30 to 35°C. The young seedlings emerged from depths of up to 9 cm with maximum emergence at 1 to 3 cm. Dorney and Wilson (1990) found that seedlings established best when left on the soil surface, particularly when covered by mulch. Seeds had no dormancy period and germinated in response to sufficient water.

A regrowth of the axillary buds of young plants is observed if a mechanical treatment or contact herbicide destroys the upper leaves. This process occurs if enough light is received (Kissman and Groth, 1993). Furthermore, Langston et al. (1984) have shown that *E. heterophylla* has an unusual and remarkable ability to regenerate by adventitious buds developing from below the cotyledonary node after shoot excision. There was 100% recovery after such excision at cotyledon and 4-leaf stages. Of 17 weed species studied, the only others to show even partial recovery were *Aeschynomene indica* and *A. virginica.*

E. heterophylla is a C4 plant and its growth habit is highly dependent on light intensity. Paliwal and Ilangovan (1988) performed autoecological studies on several species, including *E. heterophylla*, which demonstrated that photosynthetic processes and the rate of photosynthesis decreased with increasing leaf age. For *E. heterophylla*, a good correlation was evident between the photosynthetic rate, stomatal resistance, protein content, transpiration rate, biomass, photosynthetic pigments, and nitrate reductase activity.

Soybean cultivars in competition with *E. heterophylla* at three densities and two periods of occurrence were studied by Chemale and Fleck (1982) in Brazil. Soybean seed yield was reduced by weed competition; the number of pods and seeds decreasing with increasing weed density. Only the highest density reduced stem diameter and node numbers. Weed populations varied most markedly with changing seasons and different levels of fertilizer application in different crops. *E. heterophylla* was among the species most promoted by fertilizers (Marnotte, 1984).

Remison (1978) found that in the greenhouse, cowpea competition with *E. heterophylla* decreased the height of the plant, the number of nodes, peduncles, green leaves, and the weight of pods and seeds; and the decrease was greater with increasing density of the associated weed. In competition with the weed, cowpea did not respond to the application of fertilizers. In field experiments, competition from the natural weed flora affected the number of days to 50% flowering, as well as, other yield components of four cowpea cultivars studied. The yield of the climbing variety, Dinner, was least affected by competition whilst the semi-erect variety, Ife brown, was affected most.

Mohamed-Saleem and Fawusi (1983) studied the allelopathic effects of plant material of several plants, including that of *E. heterophylla*, on tomato, pepper and sorghum. They found that increasing amounts of decomposed weeds significantly reduced germination and seedling growth, although *E. heterophylla*

had the least effect. Allelopathic effects of seven weed species on pumpkin and eggplant were studied under greenhouse conditions by Almodovar-Vega et al. (1988a, b). Root exudates from the roots of several plants, including *E. heterophylla*, decreased the vine length and dry weight of pumpkin seedlings when added to their growth medium.

Pest Importance

Nester et al. (1979) contend that soybean yields can be reduced to zero by dense infestations of *E. heterophylla* (Fig. 2), and even moderate infestations can cause difficulties at harvest and reduce quality by contamination with sticky latex and adhering dirt and trash.

According to Holm et al. (1979), *E. heterophylla* is a major weed problem in Fiji, Ghana, Mexico, Philippines, Indonesia and Thailand, and a principal weed in Brazil, India, Italy, Papua New Guinea, Cuba, Honduras, Peru, Uganda and the U.S. Crops in which it is reported as a major weed include cocoa, coffee, cotton, cowpeas, maize, papaya, peanut, sorghum, soybean, sugarcane, tea and upland rice (Parsons and Cuthbertson, 1982). Because of its rapid growth, it can be a serious competitor early in the life of the crop, competing for light as well as for water and nutrients. *E. heterophylla* was planted in soybean rows at a rate of 2.5 plants per foot (approximately 8/m) and allowed to compete for 8 weeks, 12 weeks and a full season. Yields were reduced by 18, 22 and 33%, respectively (Harger and Nester, 1980).

Figure 2. *E. heterophylla* infestation. Photo courtesy of Novartis (CABI, 2004).

E. heterophylla interference with peanuts was studied by Bridges et al. (1992) in the U.S. Based on four field experiments with differing densities of *E. heterophylla*, peanut yield losses in Georgia were predicted to be 0, 4, 8, 12, 15, 26, 40 and 54% for season-long *E. heterophylla* interference at densities of 0, 1, 2, 3, 4, 8, 16 and 32 plants/9 m of row, respectively. In Florida, predicted peanut yield losses were 0, 9, 14, 22, 30, 37 and 41% for weed densities of 0, 1, 2, 4, 8, 16 and 32 plants/9 m row, respectively. Peanuts had to be maintained weed free for 10 weeks after peanut emergence to prevent yield loss. *E. heterophylla* that interfered with peanuts for more than 2 weeks after emergence of the crop reduced yields.

Symptoms/Signs

E. heterophylla is herbaceous, erect and 20 to 200 cm in height (depending on growing conditions). The most common size is 40 to 60 cm tall. Milky latex is present when most parts of the plant are broken. *E. heterophylla* has an annual, spring-summer cycle and competes with crop species to reduce yield.

Known Hosts

Major hosts

Arachis hypogaea (peanut), *Glycine max* (soybean), *Gossypium* (cotton), *Saccharum officinarum* (sugarcane), *Vigna unguiculata* (cowpea), *Zea mays* (maize)

Minor hosts

Allium cepa (onion)

Known Distribution

E. heterophylla originated in the tropical and subtropical regions of America but is now distributed throughout tropical Africa and Asia in a total of at least 65 countries. Countries with the weed present include: **Asia:** Bhutan, Cambodia, China, India, Indonesia, Israel, Japan, Jordan, Laos, Malaysia, Myanmar, Nepal, Philippines, Sri Lanka, Thailand, Vietnam; **Europe:** Italy; **Africa:** Botswana, Congo Democratic Republic, Egypt, Ethiopia, Ghana, Kenya, Mauritius, Mozambique, Nigeria, Senegal, South Africa, Sudan, Tunisia, Uganda, Zambia, Zimbabwe; **North America:** Mexico, the U.S.; **Central America:** Costa Rica, Cuba, Dominican Republic, El Salvador, Guatemala, Honduras, Jamaica, Lesser Antilles, Nicaragua, Puerto Rica, Trinidad and Tobago; **South America:** Argentina, Bolivia, Brazil, Colombia, Ecuador, Paraguay, Peru, Suriname, Venezuela; **Oceania:** Australia, Federated states of Micronesia, Fiji, and Papua New Guinea.

Potential Distribution Within the US

The weed is present in Alabama, Arizona, California, Hawaii, Florida, Georgia, Louisiana, Mississippi, New Mexico, and Texas.

Survey

E. heterophylla grows in moist tropical and subtropical regions on a wide range of soils, principally in shaded waste places and in cultivated areas (Parsons and Cuthbertson, 1982). Milky latex is present when most parts of the plant are broken.

Key Diagnostics

E. heterophylla is not readily confused with any other common weedy species, with the exception of *E. cyanthophora,* which has similar variable leaf shape but the bracts mainly red (as opposed to green in *E. heterophylla*), nectaries sub-sessile with elliptical opening (stalked with round opening in *E. heterophylla*), fruits ovoid-angular (v. trigonous) and seeds tuberculate, without caruncle

(prismatic tuberculate, with caruncle in *E. heterophylla*) (Oliviera and Sa-Haiad, 1988).

Mimosa diplotricha

Scientific Name
Mimosa diplotricha Sauvalle

Synonyms:
Mimosa invisa, Morongia pilosa, Schrankia brachycarpa, Schrankia pilosa

Common Name(s)
Giant sensitive plant, nila grass

Type of Pest
Weed

Taxonomic Position
Class: Dicotyledonae, **Order:** Fabales, **Family:** Fabaceae

Reason for Inclusion in Manual

Pest Description
In its native range, *Mimosa diplotricha* (Fig. 1) behaves as a perennial, but in its introduced range it can be an annual, biennial or perennial shrub. It is characterized by robust growth, which enables it to scramble over other vegetation, forming spreading, impenetrable, tangled thickets of undergrowth. Due to its rapid growth rate, each plant can cover an area of 2 to 3 m² in one growing season. It is extremely invasive, highly competitive, a prolific seed producer, and capable of spreading rapidly (CABI, 2004).

Flowering may occur throughout the year but is concentrated late in the wet season. In Australia, it usually flowers and seeds from April through to the end of June, but in years when there is little cold weather, plants will seed from April through to December. Some plants can set seeds when only 10 cm high.

An erect, climbing, ascending or prostrate biennial or perennial shrub that often forms a dense thicket, the root system strong, often woody at the decumbent base; stems conspicuously angular throughout the length, up to 2 m tall with many randomly scattered recurved spines or thorns 3 to 6 mm long; leaves bipinnate, 10 to 20 cm long, moderately sensitive to the touch; pinnae four to nine pairs; leaflets 12 to 30 pairs, sessile, opposite, lanceolate, acute, 6 to 12

mm long, 1.5 mm wide; inflorescence a head, one to three in the axils of leaves, on stalks 1 cm long, hairy, about 12 mm in diameter; corolla united at least at the base (gamopetalous), pale pink; stamens twice as many as the petals; fruit a pod, spiny, three- to four-seeded, borne in clusters, linear, flat, 10 to 35 mm long, splitting transversely into one-seeded sections which separate at grooves or seams (sutures); seeds flat, ovate, 2 to 2.5 mm long, light brown (Holm et al., 1977).

Up to 20,000 seeds/m²/year can be produced by *M. diplitricha*. Even seedlings a few weeks old can produce viable seed. Although the plant produces copious quantities of flowers, the percentage of floral and/or fruit abortions in Peninsular Malaysia is about 45 to 50%. Those in the north rarely produce fruits; whereas those in the south produce fruits in abundance.

Figure 1. *M. diplotricha***,** giant sensitive plant, stem and flowers. (top), close-up of flowers (bottom). Photos courtesy of Colin Wilson (CABI, 2004).

The plant is extremely persistent because it produces physically and physiologically hard seeds, which can survive in the soil for many years. Seeds may remain dormant for up to 50 years. The seeds have a long dormancy which can be broken by the heat from grass fires. The spiny seed pods are adapted to dispersal by animals and floodwaters, but seeds can also be distributed in contaminated hay, impure agricultural seed and construction materials, as well as by boats, vehicles and machinery.

Biology and Ecology

In its native range, the shrub is often found in disturbed shrub-woodland, at the edge of gallery forest and open rocky places. Low temperatures limit the species, but its tolerance limits are unclear. In Australia, reproduction is limited by cold

and Hong Kong winters may be too cold for it to become an important component of the local vegetation. It is a lowland species, and in Bolivia, it has been recorded at an altitude of 270 m and up to 1000 m in Sao Paulo, Brazil (CABI, 2004).

Pest Importance

M. diplotricha is considered one of the top hundred worst weeds of the world. It is listed as a weed of 13 crops in 18 countries. It a serious weed in the Pacific islands, South-East Asia, Mauritius, and Nigeria. It rapidly smothers crops and pastures in tropical and subtropical countries, reducing yields. Where hand harvesting of crops is carried out, infested fields are made difficult and dangerous to work; the thorns can cause serious sores on humans. Mechanical harvesters can also be jammed when used in infested crops. In Nigerian cassava fields, increasing populations of *M. diplotricha* rapidly decrease cassava tuber yields. When *M. diplotricha* density reached 630,000 plants per hectare, cassava root yield 12 months after planting was reduced by 80% (CABI, 2004).

Infestations of *M. diplotricha* can be encouraged by overgrazing, thus animals are prevented from grazing in heavily infested areas. *M. diplotricha* thickets become a serious fire hazard when dry. In Papua New Guinea, *M. diplotricha* has a direct negative impact on growth, yield and harvesting of sugarcane, but no direct assessment of the actual economic losses has been made. However, on cattle ranches in the Markham Valley, up to US $130,000 is spent annually on chemical control.

There is evidence that M*. diplotricha* is toxic to livestock. In Thailand, 22 swamp buffaloes died 18 to 36 hours after eating *M. diplotricha* var. *inermis*. The symptoms were salivation, stiffness, lack of mastication, muscular tremor, dyspnea and recumbency. The toxic elements were found to be cyanide and nitrite. It is reported that a clinical case of *M. diplotricha* var. *inermis* resulted in poisoning of a 2-year-old Jersey-cross heifer in India. The severity of the clinical signs and lesions correlated well with the quantity of the weed consumed. Other animals grazing in the same area did not develop any clinical signs of toxicity, and it appears as if the toxicity is also related to the stage of growth of the plant, and various other animal factors such as the development of tolerance. Tests in Queensland, Australia, show this variety to be toxic to sheep, and a report from Flores, Indonesia, suggests that it is toxic to pigs (CABI, 2004).

M. diplotricha can out compete native plant regeneration, as well as detrimentally affect humans. The numerous sharp recurved prickles associated with a scrambling habit may give the impression, both visual and tactile, of a sort of 'organic or green barbed wire'. In Australia, it is considered to exert competition by forming dense mats, which adversely affect the growth of a number of native species. If allowed to spread in Western Australia, *M. diplotricha* is predicted to cause serious ecological impacts.

M. diplotricha is the principal weed of rubber and coconut in Papua New Guinea, rubber in Indonesia, sugarcane in Taiwan and the Philippines, lychee in Thailand, and tomato in the Philippines. It is considered a weed of sugarcane in Australia and India; cassava, soybeans, maize, apple, citrus and tea in Indonesia; coconut in Sri Lanka; rubber in Malaysia; banana and tea in India; and abaca (*Musa textilis)* and pineapple in the Philippines. It is considered a major threat to tropical pastures in Australia, the Pacific islands, Papua New Guinea and the Philippines. It is a weed of lowland rice in Indonesia, the Philippines, Thailand and Vietnam; of dry-seeded rice in the Philippines; and of upland rice in Indonesia, Laos, the Philippines, Thailand and Vietnam. It is potentially the worst weed in plantations and arable lands of Fiji and the Philippines.

Symptoms/Signs

M. diplotricha has the ability to climb over other plants and can probably shade out light-demanding species. It has been shown to reduce yield in a number of economically important crop species.

Known Hosts

Major hosts

Cocos nucifera (coconut), *Hevea brasiliensis* (rubber), *Litchi chinensis* (lichi), *Lycopersicon esculentum* (tomato), and *Saccharum officinarum* (sugarcane)

Minor hosts

Ananas comosus (pineapple), *Areca catechu* (betelnut palm), *Camellia sinensis* (tea), *Citrus, Coffea arabica* (arabica coffee), *Glycine max* (soybean), *Malus* (ornamental species apple), *Manihot esculenta* (cassava), *Musa* (banana), *Musa textilis* (manila hemp), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), and *Zea mays* (maize)

Known Distribution

M. diplotricha is native to the neotropics, including much of South and Central America, as well as the Carribean. However, it is unclear whether it is native to North America and parts of the Caribbean. It has now become widespread throughout the wet tropics and subtropics. Usually it is a very invasive species wherever introduced. Countries with the weed present include: **Asia:** Cambodia, China, India, Indonesia, Laos, Malaysia, Myanmar, Philippines, Singapore, Sri Lanka, Thailand, Timor-Leste, Vietnam; **Africa:** Burundi, Cameroon, Congo Democratic Republic, Ivory Coast, Ethiopia, Ghana, Guinea, Mauritius, Mozambique, Nigeria, Reunion, Rwanda, Tanzania, Togo, Zimbabwe; **North America:** Mexico, the U.S.; **Central America:** Costa Rica, Cuba, Guatemala, Haiti, Honduras, Jamaica, Puerto Rico, the U.S. Virgin Islands; **South America:** Argentina, Bolivia, Brazil, Colombia, Ecuador, Paraguay, Peru, Venezuela; **Oceania:** American Samoa, Australia, Belau, Cook Islands, Federated states of Micronesia, Fiji, French Polynesia, Guam, New Caledonia, Niue, Northern Mariana Islands, Papua New Guinea, Samoa, Solomon Islands, Vanuatu, Wallis and Futina (CABI, 2004).

Potential Distribution Within the US

M. diplotricha is present in Hawaii, Puerto Rico, and the U.S. Virgin Islands.

Survey

A major weed in pastures, plantations and roadsides and can also be serious in crops. It grows best where fertility, soil and air humidity, and light are all high and dies away in prolonged dry seasons.

Key Diagnostics

M. diplotricha can be easily distinguished from *M. pudica*, as *M. diplotricha* is larger and is only slightly sensitive to touch. A spineless form is occasionally found but is suspected of reverting to the spiny form.

Pharbitis nil

Scientific Name
Pharbitis nil

Synonyms:
Ipomoea nil, Convolvulus nil, Ipomoea hederacea, Ipomoea longicuspis, Ipomoea triloba

Common Name(s)
Japanese morning glory, little bell

Type of Pest
Weed

Taxonomic Position
Class: Dicotyledonae, **Order:** Solanales, **Family:** Convolvulaceae

Reason for Inclusion in Manual

Pest Description
Pharbitis nil (Fig. 1) is an annual herb with twining stems, 1 to 3 m long, glabrous except the inflorescence; stems somewhat angled, about 3 mm thick, milky, leaves broadly ovate to orbicular in outline, entire, coarsely dentate to more or less deeply 3-lobed, center lobe may be pointed, base broadly cordate, 4 to 11

Figure 1. *Pharbitis nil* leaves and flower (leaf), close-up of flower right) Photo courtesy of USDA-APHIS archives, 1715Hwww.invasive.org.

cm long, often nearly as wide; petiole slender, 3 to 10 cm, glabrous or sometimes minutely tuberculate; inflorescence axillary; peduncle shorter to longer than the petiole, angular, minutely verrucose toward the apex, one-flowered or cymosely

few to several-flowered; branched of the cyme very short; flowers aggregate; pedicels minutely verrucose or glabrous, 2.5 to 8 mm, closing before noon; sepals slightly unequal, 7 to 8 mm long, the outer ones a little shorter, oblong to narrowly elliptic-oblong, glabrous or sparsely hairy on the back with distinctly fimbriate margins; corolla is 5-lobed funnel-shaped, 1.5 cm long, glabrous, color variable around the world, pink or pale red to purple in the Philippines, and in Costa Rica may be pale red to violet, or white with a deep red-violet throat with contrasting white stamens and stigmas; stamens inserted in tube of corolla; filaments hairy at the base; ovary 2 to 4 celled, conical, densely pubescent; ovules 2 to 4; fruit a capsule, depressed globose with sharp point, bristly hairy; seeds 4 or less, 3.5 mm long, diameter 6 mm, hard, shiny, and brown.

Biology and Ecology
P. nil reproduces from seed, is self-fertile, and can produce about 180 seeds per plant. Scarified seeds in soil with 40 to 80% soil moisture will germinate. Germination reached 50% in moist sand in the Philippines. In Brazil, *P. nil* prefers crops grown in sandy soils. In Costa Rica, the plants flower in mid-October and continue through December.

Pest Importance
The plant can serve as a reservoir for Cucumber mosaic virus, *Cylas formicarius* (sweet potato weevil), *Phytophthora infestans* (Phytophthora blight), and sweet potato little leaf phytoplasma (sweet potato witches' broom).

In Indonesia, *P. nil* reduces sugar cane stalk numbers and yields and is considered one of the four main weeds in this crop. In upland rice crops in the Philippines, *P. nil* can germinate 4 days after rice planting and can eventually smother the crop. In Australia, seed germination occurs in flushes after heavy rain periods. Sugarcane crops grow too tall for season long cultivation of *P. nil*, which allows the vine to entangle the crop and jam up the mechanical harvesters during the harvest.

Symptoms/Signs
Competes with crops for nutrients and resources and reduces crop yield.

Known Hosts
Major hosts
Maize, peanuts, upland rice, soybean, sorghum, tobacco, bananas, coffee, potatoes, dry beans, cotton, and sugarcane

Known Distribution

P. nil is found on all seven continents, and occurs in 40 different crops. It is considered a weed in 40 countries. The primary range is within ±15 C isotherms north and south of the equator. It is most common in Central America, the Caribbean, south and east Asia, and Australia.

The weed is present in Brazil, the Philippines, Indonesia, Honduras, Jamaica, Australia, Nicaragua, Costa Rica, Argentina, Mexico, Bangladesh, India, Cambodia, Colombia, Cuba, Ivory Coast, Laos, Nepal, New Guinea, San Salvador, Samoa, Senegal, Thailand, the U.S., and Venezuela.

Potential Distribution Within the US

The weed is present in California, Arizona, Florida, and Hawaii.

Survey

Distinguishing features of the plant are 3-lobed leaves, stems with milky sap, and the hirsute or hairy ovary. In Brazil, stems have white to transparent hairs.

Mollusks

Cernuella virgata

Scientific names
Cernuella virgata Da Costa

Synonyms:
Cernuella virgatus, *Cernuella variabilis, Cernuella virgata* ssp. *variegate*, *Helicella maritime*, *Helicella variabilis*, *Helicella virgata*, *Helix virgata*

Common Name(s)
Maritime garden snail, Mediterranean snail, Mediterranean white snail, striped snail, vineyard snail, white snail

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Stylommatophora (Eupulmonata), **Family:** Hygromiidae (Helicidae)

Reason for Inclusion in Manual

Pest Description
The shell of *C. virgata* is globose-depressed, and white or yellowish-white in color with dark-brown bands or spots (Fig. 1, 2). Snail size is 6 to 19 mm high x 8 to 25 mm wide (GPDD). Shell size and banding patterns are reported to vary widely geographically throughout Southeastern Australia

Figure 1. Banding of *C. virgata*. Photo courtesy of Tenby Museum.

(Baker, 1988b). Size has been demonstrated as inversely proportional to population density (Baker, 1988b). *C. virgata* is considered polymorphic; banded and unbanded (more common) morphs have been found throughout Australia. Relative frequencies of each morph are likely correlated with site-specific factors such as predator pressure (Baker, 1988b).

Figure 2. *C. virgata*. Photo courtesy of L. Poggiani. 1716Hwww.lavalledelmetauro.it

The Mediterranean snail is relatively small and is characterized by prominent spiral banding on the shell (Fig. 1).

Biology and Ecology

C. virgata has an annual life cycle; breeding occurs from fall through winter. Snail breeding beings at the onset of post-summer rains in South Australia. Five phases are distinguished in the terrestrial gastropod reproductive cycle: courtship and copulation occur on the surface of the soil, while nest-building, egg-laying and embryonic development, and hatching of young occur in the soil. Based on laboratory observations, snails are thought to spend considerable time on the soil surface during mating and oviposition. Mating by *C. virgata* is usually observed following rain. One to two hundred eggs are typically laid if rain continues after mating and the majority of adults die following reproduction. In the laboratory, breeding pairs of snails produced 50 to 8268 eggs when provided with unlimited food (Charwat et. al., 2000).

Immature snails can be differentiated from adults based on 1) initially smaller size; and 2) lack of rib formation on the inside of shells in early autumn (Baker, 1988b).

This snail typically aestivates atop various structures (Fig. 3) such as plants and fence posts (OSU, 2004). *C. virgata* will also climb onto and feed upon new growth, causing

Figure 3. Multiple *C. virgata* on tree trunk. Photo courtesy of L. Poggiani, 1705Hwww.lavalledelmetauro.it

significant damage. When extreme hot or cold temperatures arrive, they withdraw into their shells and seal the opening with an epiphragm (a thin layer of hardened mucus and calcium). This tactic (termed aestivation during hot periods, and hibernation or diapause during cold periods) allows the snails to exist in a state of suspended metabolic activity until more favorable warm, moist conditions arise. *C. virgata* is thought to be nocturnal with its activity closely linked to moisture availability (GPDD).

C. virgata movements are reported as between 0.1 and 0.4 meters per day. Movement of more than 25 meters in one month during spring and summer and 50 meters in three months in autumn and winter was recorded in Australia (Baker, 1988a).

C. virgata are hermaphrodites and obligate outcrossers (Charwat et al*.,* 2000).

Pest Importance

Introduced Mediterranean snails are serious agricultural pests in South and Western Australia, among other regions. They can cause severe damage to and, at times, destroy legume and seedling crops (CSIRO). *C. virgata* was first found in Western Australia in 1984 (Baker, 1988b). *C. virgata* can prompt significant economic crop losses, on grains, in particular. These snails aestivate on plant heads and stalks, which contaminate crops and clog machinery (Baker, 1988b; Coupland, 1995; OSU, 2004). Areas previously infested with snails can prevent re-establishment of site as pastureland, as livestock often reject slime-contaminated hay and forage (GPDD).

It is thought that *C. virgata* principally feeds on decayed organic material (Baker, 1988b). Several congeners of *C. virgata* are also considered to be highly damaging pests.

Control of *C. virgata* may be achieved with cultural, mechanical, chemical, and biological control. Metaldehyde baits are commonly used to kill snail pests (Baker et al*.,* 2004). Several nematode parasites are available or under development currently. One, *Phasmarhabditis hermaphrodita*, has recently been made available commercially in the United Kingdom. Large numbers of this nematode were shown to be necessary for mortality of non-breeding snails to occur in laboratory soil-based assays (Charwat et al., 2000).

Symptoms/Signs

C. virgata is found atop plants during summertime (Fig. 3) and may also be found feeding on new growth earlier in the season. These snails aestivate on plant heads and stalks, which contaminate crops and clog machinery. Areas previously infested with snails can prevent re-establishment of the site as pastureland, as livestock often reject slime-contaminated hay and forage.

Known Hosts

Major Hosts
Brassica napus (canola), *Glycine max* (soybean), *Gramineae* spp. (cereals), *Helianthus annuus* (sunflower), *Hordeum* spp. (barley), *Medicago sativa* (Alfalfa), *Medicago* spp. (medic), *Micropus* spp. (cottonseed), *Pisum* spp. (pea), *Poaceae* spp. (grasses), *Trifolium* spp. (clovers), *Triticum* spp. (wheat), and Vitaceae (grapes).

Known Distribution
Known to occur in Albania, Australia, Austria, England, New Zealand and United Kingdom.

Potential Distribution Within the US
C. virgata is known to occur in Washington and may easily adapt to the mild climates of the Pacific Northwest.

Survey
C. virgata is a conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight, or in the morning or evenings following a rain event. *C. virgata* is easily seen, and attacked plants exhibit extensive rasping and defoliation. Like other mollusks, it can also be detected by signs of ribbon-like excrement and slime trails on plants and buildings.

Key Diagnostics
C. virgata is a relatively small snail (up to 15mm in diameter) characterized by prominent spiral banding on the shell.

C. virgata closely resembles the white Italian snail (*Theba pisana*) in appearance and pest status (GPDD). *C. virgata* can be differentiated from *T. pisana* based on more pronounced spiral banding and the umbilicus (hole about which the shell spirals) appears as a circular hole rather than being partially obscured as in the white Italian snail. *T. pisana* is also established in California.

Glossary

Abaxial: Concerning the surface of a structure that is turned away from the structure's primary axis, pertaining to the lower surface of a leaf.

Abdomen: The posterior of the 3 main body divisions of an insect. Bears no functional legs in the adult stage.

Acervulus (pl. acervuli): Erumpent, cushionlike fruiting body of a fungus bearing conidiophores, conidia, and sometimes setae.

Acicular: Needle-shaped.

Acrostichal setae/hairs: Very short hairs between the dorsocentral bristles.

Acuminate: Gradually narrowing to a point.

Adaxial: Located on the side or directed toward the axis, pertaining to the upper surface of a leaf.

Adventitious roots: Root growing in an unusual location e.g. from a stem.

Aedeagus: In male insects, the penis or intermittent organ, situated below the scaphium and enclosed in a sheath.

Aerobic: Living only in the presence of oxygen.

Aestivate: To pass the summer in a dormant or torpid state.

Agglutinate: To cause to adhere, as with glue.

Alae: Expansions or projections formed by a longitudinal thickening of the cuticle of a nematode. Cervical alae are confined to the anterior region of nematodes parasitic in animals. Caudal alae occur in the posterior region of males in a number of genera. Longitudinal alae, usually four, extend the length of the body sub laterally.

Alatae: Winged forms of aphids or aphids with wings.

Amphid: A chemosensory organ located in the anterior region of a nematode.

Amphidelphic: Nematodes: having two ovaries, one directed anteriorly and the other posteriorly.

Amphigenous: Growing all around.

Anabolism: The metabolic synthesis of complex molecules from simpler ones.

Anal: In the direction or position of the anus, near the anus or on the last abdominal segment.

Anal hooks: Lepidoptera pupae: hooked or clubbed setae at the apex of the abdomen that attach the pupa to the cocoon or silk pad.

Anal lobe: 1) Hymenoptera: the posterior lobe of the wings; 2) Diptera: the basal part of the wing behind the anal vein; 3) Coccidae: a pair of small, triangular, hinged processes forming a valve that covers the anal orifice; 4) Immatures: any protrusion of the integument near the anus.

Anal plate: 1) Lepidoptera larvae: The shield–like covering of the dorsum of the last segment; 2) Embryonic larvae: tergum XI, 3)Cocciids: a pair of triangular or semicircular sclerites at the cephalic end of the caudal cleft.

Anamorph: The asexual form in the life cycle of a fungus, when asexual spores (such as conidia) or no spores are produced.

Anhydrobiosis: A metabolic state of life entered by some lower organisms in response to adverse dessication conditions.

Annule: Thickened interval between transverse striae in the cuticle of a nematode.

Annellus: Hymenoptera: small ring-like segments between the pedicel and funicle of the antenna.

Antenna (pl. Antennae): One of the paired segmented sensory organs borne one on each side of the head, maybe referred to as horns or feelers.

Antennal club: A variable number of segments of the antennal flagellum usually identified by a change in shape or form from preceding segments. The antennal club is always apical, is sometimes arbitrarily delimited by segment number and always includes the terminal segment.

Anterior: In front, before.

Anther: Pollen-bearing portion of a flower.

Antheridium (pl. antheridia): A male gametangium.

Anthesis: The period of the opening of a flower during which pollination can occur.

Anthracnose: Disease caused by acervuli-forming fungi (order Melanconiales) and characterized by sunken lesions and necrosis.

Aperture (pl. Aperatures): To uncover, to open. Any opening in a wall, surface or tube.

Apical: At, near, or pertaining to the apex of any structure.

Appresorium (pl. Appressoria): Swollen, flattened portion of a fungal filament that adheres to the surface of a higher plant, providing anchorage for invasion by a fungus.

Apterae (pl. aptera): Insects that have no wings.

Arcuate: Curved like a bow.

Arista: A large bristle, usually located on the apical antennal segment (Diptera).

Arthropod: Any of numerous invertebrate animals of the phylum Arthropoda, including the insects, crustaceans, arachnids, and myriapods, that are characterized by a chitinous exoskeleton and a segmented body to which jointed appendages are articulated in pairs.

Ascus (pl. asci): Saclike structure containing ascospores (typically eight) and usually borne in a fungal fruiting body.

Ascoma (pl. ascomata): Sexual fruiting body of an ascomycetous fungus that produces asci and ascospores; e.g. apothecium, ascostroma, cleistothecium, perithecium, pseudothecium.

Autoecious: In reference to rust fungi, producing all spore forms on one species of host plant.

Axil: The angle formed by the leaf petiole and the stem.

Axillary: Pertaining to or placed within an axil.

Axillary bud: Bud that develops in the axil of a leaf.

Awn: Bristle-like structure at the apex of the outer bract of some cereal and grass flowers.

Basal: Pertaining to the base or point of attachment to or nearest the body.

Biguttinate: With blunt ends.

Biotype: A group of organisms having the same genotypes but may vary in biological and phenological differences.

Blight: Sudden, severe, and extensive spotting, discoloration, wilting, or destruction of leaves, flowers, stems, or entire plants.

Bulb: Any pear-shaped protuberance or structure that resembles a bulb.

Bursa: Nematodes: Caudal alae of males used to clasp the female during copulation.

Callus (pl. calli): 1) A hard lump or mound-like, rounded swelling of the integument, such as a swelling at the base of the wing articulating with the thorax; 2) Heteroptera: the thickened or raised spots on the thorax, especially of Pentatomidae.

Calyx: The outer-most group of leaves surrounding the flower; the external-most part of the flower.

Capsomere: Protein subunits that serve as components of the viral capsid.

Carlavirus: (Siglum of carnation latent virus.) Member of a group of plant viruses with slightly flexuous, rod-shaped particles containing a single molecule of linear RNA, most of which are transmitted by aphids in a noncirculative manner.

Caruncle: A localized outgrowth or appendage of the seed coat near the hilum of a seed.

Caudate: 1) With tail-like extensions or processes; 2) Hymenoptera (Apocrita): specialized body form of some endoparasitic ichenumonid larvae, characteristically segmented, with long, flexible, caudal appendages. Function of cadual appendages not established, but sometimes progressively reduced in later instars and lost in the last instar.

Caudal: At or towards the anal (tail) end.

Cell wall: Protective, resistant, but permeable structure secreted externally to the cell membrane in plants, bacteria, fungi, and certain other organisms.

Cephalic: Pertaining to the head.

Cephalopharyngeal apparatus: Larval feeding organ consisting of mouth hooks, H Piece, medial tooth and cephalopharyngeal skeleton.

Chlamydospore: Thick-walled or double-walled asexual resting spore formed from hyphal cells (terminal or intercalary) or by transformation of conidial cells that can function as an overwintering stage.

Chlorosis (adj. chlorotic): Failure of chlorophyll development, caused by disease or a nutritional disturbance; fading of green plant color to light green, yellow, or white.

Chord: Nematodes: a longitudinal internal thickening of the hypodermis.

Chorotic: Abnormal condition of plants in which the green parts lose their color or turn yellow as a result of chlorophyll production due to disease or lack of light.

Cilium (pl. cilia): Fringes arranged from series of moderate or thin setae arranged in tufts or single lines; thin scattered setae on a surface or margin.

Cinereous: Possessing the qualities of ash-colored, grey tinged with black.

Clavate: Club-shaped.

Cleistogamous: Flowers that do not open and are self pollinated. Cleistogamy insures that a plant produces seeds, even if conditions are unfavorable for wind or insect pollination.

Cloaca: Nematodes: a common duct or cavity in which the digestive and reproductive systems terminate in males.

Coat protein: The protective layer of protein surrounding the nucleic acid core of a virus; the protein molecules which make up this layer.

Coecum: A blind sac or tube. Term applied to a series of appendages opening into the alimentary canal at the junction of a crop and chylific ventricle.

Complete metamorphosis: Metamorphosis in the holometabola which has four stages: egg, larva, pupa and adult. Each stage entirely different from the others.

Conidioma (pl. conidiomata): Specialized conidia-bearing structure, e.g. acervulus, pycnidium, sporodochium, synnema.

Chorion: The outer shell or covering of the insect egg.

Comovirus: (Siglum of cowpea mosaic virus). Member of a group of multicomponent plant viruses with small, isometric particles containing two linear RNA species, readily transmitted mechanically and by beetles.

Conidioma (pl. conidiomata): Specialized conidia-bearing structure, e.g. acervulus, pycnidium, sporodochium, synnema.

Conidiophores: Simple or branched hypha on which conidia are produced.

Conidium (pl. conidia): An asexual, nonmotile fungal spore that develops externally or is liberated from the cell that formed it.

Cornicle: A pair of tubes on the abdomen of aphids that discharge defensive secretions, especially alarm pheromones (also siphunculi).

Corolla: Petals, collectively.

Corpus bursae: The sac-like portion of the bursa copulatrix which bears the ostium bursae.

Cortex (adj. cortical): The portion of a stem or root that is external of the vascular tissue.

Costa: 1) An elevated ridge that is rounded at its crest; 2) the thickened anterior margin of a wing, typically referring to the forewing; 3) the vein extending along the anterior margin of the wing from base to the point of junction with the subcosta.

Cotyledon: A leaf of the embryo of a seed plant, which upon germination either remains in the seed or emerges, enlarges, and becomes green; also called seed leaf.

Cremaster: 1) The apex of the last segment of the abdomen; 2) the terminal spine or hooked process of the abdomen of subterranean pupa. Used to facilitate emergence from the earth; 3) an anal hook by which some pupae are suspended.

Crochets: Tiny hooks on the prolegs of caterpillars.

Crustose: A crust-like growth form that is closely attached to the substrate.

Ctenidium (pl. ctenidia): A comb-like row of short non-innervated spines (bristles) on an insect's body. The rearward orientation of the spines is believed to facilitate movement among hairs on the host's body.

Cucullus: 1) A hood, a hood-shaped covering or structure; 2) genitalia of male Lepidoptera: terminal part of the harpe.

Cultivar: A plant type within a species, resulting from deliberate genetic manipulation, which has recognizable characteristics (color, shape of flowers, fruits, seeds and height or form).

Cuneate: Wedge-shaped; descriptive of structure that is elongate-triangular. Term often applied to leaves with abruptly pointed apex and tapering to the base.

Cuticle: The noncellular outer layer of the body wall of an arthropod.

Cuticula: The outer body wall of an insect; “thin skin”.

Dehiscent: 1) Botany: splitting open at maturity to release contents (of a fruit); 2) Pathology: of an ascus or fruit-body, opening when mature, by a pore or by rupturing or fragmentation; of conidia and other spores, falling off.

Denticles: A small tooth or cuticular projection.

Detritus: Material which remains after disintegration; rubbing away or the destruction of structure; fragmented material; any disintegrated or broken matter.

Deutonymph: The third instar of a mite.

Diapause: A condition of restrained development and reduced metabolic activity, which cannot be directly attributed to unfavorable environmental conditions. Regarded by entomologists to involve a resting period of an insect, especially of larvae in winter. (hibernation, quiescence).

Dichromatism: The expression of two color forms (morphs) within on species.

Dicotyledonous (dicots): A flowering plant with two seed leaves characterized by embryos with two cotyledons, net veined leaves, flower parts in fours or fives.

Dimorphism: A genetically controlled, non-pathological condition in which individuals of a species are characterized by distinctive or discrete patterns of coloration, size or shape. Dimorphism can be a seasonal, sexual or geographic manifestation.

Direct penetration: Penetration of plant tissues by a pathogen through barriers such as leaf cuticle by chemical and physical means (e.g. penetration peg).

Discal: On or relating to the disc of any surface or structure.

Diurnal: 1) Relating to or occurring in a 24-hour period; daily; 2) occurring or active during the daytime rather than at night: diurnal animals; 3) Botany: opening during daylight hours and closing at night.

Diverticulum: A tube, sac or invagination originating on the wall of a vessel or the alimentary canal and closed at the distal end.

Doliiform: Barrel-shaped to broadly subglobose.

Dorsal: On the upper surface.

Ductus bursae: The duct in female Lepidoptera extending from the ostium to the bursa copulatrix.

Ductus seminalis: Female Lepidoptera: the tube or canal connecting the bursa copulatrix with the common oviduct.

Ecdyses: Molting; the process of shedding the exoskeleton.

Echinulate: Having small spines projecting from cell walls.

Eclosion: Hatching from the egg.

Elytra: The anterior leathery or chitinous wings of beetles.

Embryo: An organism in the early stages of development, such as a young plant in the seed, or a nematode before hatching from the egg.

Encapsidate: To cover virus nucleic acid with a protein coat.

Envelope: Virology: a protein covering that packages the virus's genetic information.

Epidermis: The cellular layer of the skin; secreting the cuticula of insects.

Epiphragm: A thin layer of hardened mucus and calcium.

Epiphytic: Living on the surface of plants but not as a parasite.

Epitope: An amino acid (or other) sequence that effects formation of an antibody.

Epicraneal suture: A Y-shaped line of weakness on the vertex of the head where the split at molting occurs.

Erumpent: Bursting or erupting through the substrate surface.

Exocarp: The outer layer of the pericarp.

Exoskeleton: The entire body wall, to the inner side of which muscles are attached; the outside skeleton in insects.

Exuviae: The cast skin of an arthropod.

Facultative diapause: May or may not need to diapause; not required for development.

Feces: Excrement; the eliminated wastes of the digestive process.

Falcate: (Of spores) sickle-shaped.

Fascia: 1) Anatomy: a thin layer of connective tissue that covers, supports, binds, or connects muscles or body organs; 2) Taxonomy: a transverse band or broad line.

Fascicle: Small group, bundle, or cluster.

Femur (pl. femora): The third and usually the stoutest segment of the insect leg. Articulated with the body via the trochanter and bearing the tibia at its distal margin.

Filament: The part of the stamen which supports the anther. The stalk of the stamen.

Flange: 1) A projecting rim or edge that provides structure support and mechanical strength; 2) a part that spreads out like a rim.

Floccose: Having a cottony appearance.

Foot cell: The base of the conidiophore, where it merges with the hyphae, giving the impression of a foot; typically seen in *Aspergillus* spp.

Forewing: The anterior wing of an insect which is attached to the mesothorax.

Frass: Plant fragments made by a wood-boring insect usually mixed with excrement; solid larval insect excrement.

Frons: The head sclerite bounded by the frontal (or frontogenal) and epistomal sutures and including the median ocellus.

Frontal suture: 1) The suture between the front and the clypeaus; 2) Diptera: separates the frontal lunule from the part of the head above it; 3) Coleoptera: clypeal suture or the suture formed by the arms of the epicranial suture.

Fundatrigenia: Aphididae: Apterous, viviparous, parthenogenetic females which live on primary host plants. Progeny of the fundatrix.

Fundatrix (pl. fundatrices): Aphididae: apterous, viviparous, parthenogenetic females that emerge during spring from overwintered eggs. Morphologically, fundatrices are characterized by smaller eyes, legs and other body parts.

Funiculus: 1) Pathology: a fine rope of hyphae; the cord attaching the peridiole to the inner wall of a basidioma in some Nidulariales; 2) Botany: the stalk of an ovule.

Fuscous: Of, or pertaining to dark brown, approaching black; a plain mixture of black and red.

Fusiform (syn. fusoid): Spindle-shaped; tapering at each end.

Gastropod: Any of a large class (Gastropoda) of mollusks, usually with a univalve shell or no shell and a distinct head bearing sensory organs, such as snails and slugs.

Genu (pl. genua): A knee; the articulation between femur and tibia.

Geotrophism: Reaction towards the earth or ground.

Geminate: Situated in pairs; bifoliolate, a leaf with 2 leaflets arising from the same point.

Geniculate: Abruptly bent or twisted.

Genome: The genetic information for an organism, consisting (in the case of viruses) of one or more species of either RNA or DNA, but not both.

Germ Pore: An unthickened spot in a spore or conidial wall through which a germ tube may form.

Germ Tube: Hypha resulting from an outgrowth of the spore wall and cytoplasm after germination.

Glabrous: Without hairs.

Globose: Descriptive of structure which is spherical or globular in shape.

Glume: One of two bracts at the base of a grass spikelet.

Grub: An elongate, whitish insect larva. The term is loosely applied to all insects, but more specifically applied to the larvae of Coleoptera, and some Hymenoptera.

Gubernaculum: Nematodes: Spicule guide; sclerotized accessory piece.

Gynopara: Special parthenogenic females that produce sexual females.

Hemocoel: The cavity in which most of the major organs of the arthropod body are found, it is filled with the fluid hemolymph (the arthropod equivalent of blood), which is pumped by a heart and which circulates among the organs directly without the use of capillaries.

Hamate: Hooked; bent at the end into a hook.

Hastiseta (pl. hastisetae): One of three setal types found on the body of larval Dermestidae. Typically these are filiform, barbed and spear-tipped in form. Prone on the body when the larva is calm, but become erect when the larva is irritated.

Haulm: The part of a plant above ground, usually after the crops have been gathered or the plant is dying.

Haustorium (pl. haustoria): Specialized branch of a parasite formed inside host cells to absorb nutrients.

Hemiparasitic: Obtaining water and nutrients from the roots of other plants then manufacturing food through photosynthesis.

Hemizonid: Nematodes: lens-like structure situated between the cuticle and hypodermal layer on the ventral side of the body just anterior to the excretory pore; generally believed to be associated with the nervous system.

Hermaphrodite: An anomalous condition in humans and animals in which both male and female reproductive organs and secondary sexual characteristics are present in the same individual. The presence of both male and female reproductive organs in a plant or animal, as in an earthworm or a monoecious plant.

Heteroecious: Pertaining to a rust fungus requiring two unrelated host plants for completion of its life cycle.

Hilum: Seed scar where the funiculus was once attached. Also may designate the central part of a starch grain.

Hilar: Of or relating to or located near a hilum.

Hindwing: The posterior wing of an insect, attached to the metathorax.

Holocyclic: Having a complete life cycle. Refers to those aphids which alternate parthenogenetic with sexual reproduction, thus starting another cycle by laying winter eggs.

Homothallism (adj. homothallic): Condition in which sexual reproduction occurs with a single thallus; self-fertile.

Host cell: A cell that is infected by a virus or another type of microorganism.

Hull: Dry outer covering of a fruit or seed or nut.

Humeral: Pertaining to the shoulder; located in the anterior basal portion of the wing.

Humeral callus: Diptera: each of the anterior angles of the prescutum of the mesothorax, usually a more-or-less rounded tubercle.

Hyaline: Like glass, transparent colorless.

Hymenium (pl. hymenia): A palisade-like layer of asci or basidia, including any sterile cells; continuous, spore-bearing layer of a fungus fruiting body.

Hyperplasia: Abnormal increase in the number of cells, often resulting in the formation of galls or tumors.

Hypertrophy (adj. hypertrophic): Abnormal increase in the size of cells in a tissue or organ, often resulting in the formation of galls or tumors.

Hypocotyl: The part of the axis of a plant embryo or seedling plant that is below the cotyledons.

Hypophyllous: On the underside of a leaf surface.

Imago: The adult stage or sexually mature insect.

Immunogen: A substance capable of provoking an immune response. Also called an antigen.

Incisure: Nematodes: A longitudinal cuticular cleft that divides the lateral fields; sometimes called involution or line.

Inclusion body: Structure developed within a plant cell as a result of infection by a virus, often useful in identifying the virus.

Incubation Period: The time between penetration of a host by a pathogen and the first appearance of disease symptoms; the time during which microorganisms inoculated onto a medium are allowed to grow.

Inoculate: 1) To communicate a disease to (a living organism) by transferring its causative agent into the organism; 2) to implant microorganisms or infectious material into (a culture medium).

Inoculum: Pathogen or its parts, capable of causing infection when transferred to a favorable location.

Instar: The period or stage between molts in the larva, numbered to designate the various periods; e.g., the first instar is the stage between the egg and the first molt.

Integument: The outer covering or cuticle of the insect body.

Intercalary: Formed or situated somewhere between apex and base of a given structure.

Interspaces: 1) Coleoptera: the plane surface between elytral striae; 2) Lepidoptera: spaces between wing veins not included in closed cells; 3) Orthoptera: a deep incision or sulcus on the posterior margin of the metasternum.

Isometric: Usually used for virus particles to describe those that are icosahedral in structure and appear approximately round.

Jowls: Diptera: the cheeks behind the depressed anterior part.

Juxta: Male Lepidoptera: a sclerite beneath the aedeagus and to which it may be hinged or fused; part of the fultura inferior.

Labrum: The 'upper lip', forming the roof of the preoral cavity and mouth; derived from the first head segment.

Lamella: A thin plate or leaf-like process; a parademe.

Lamina: A thin, flat, chitinous scale-like sclerite.

Lanceolate: Spear-shaped, gradually tapering toward the extremity.

Larvae (pl. for larva): An early, free-living immature form of any animal that changes structurally when it becomes an adult usually by complex metamorphosis.

Latent Period: The time between infection and the production of new inoculum; the time after a vector has acquired a pathogen and before it can be transmitted.

Leaf spot: A plant disease lesion typically restricted in development in the leaf after reaching a characteristic size.

Lemma: The lower of two bracts enclosing the flower in grasses.

Lenticel: Corky structure on young growth that allows passage of air into the twig or trunk; an opening, usually characterized as an eruption of the periderm through which gaseous exchange may occur in stems.

Lesions: Localized diseased area or wound.

Leukocytes: Any of the various white blood cells that together make up the immune system. Neutrophils, lymphocytes, and monocytes are all leukocytes.

Litura: An indistinct spot with pale margins; a spot which appears blotted.

Locule: A cavity within which specialized organs may develop, most usually the ovules or pollen grains.

Lodge: To fall over.

Luteovirus: Literally "yellowish". Member of a group of plant viruses with isometric particles containing one molecule of linear RNA, mainly confined to the phloem, and usually not mechanically transmitted but transmitted in nature by aphids in a circulative manner.

Lysis: Rupture or destruction of a cell.

Macroconidium (pl. macroconidia): The larger of two kinds of conidia formed by certain fungi.

Mandible: The first pair of jaws in insects, stout and tooth-like in chewing insects, needle or sword-shaped in piercing-mouthed sucking insects; the lateral upper jaws of biting insects; in muscoid larvae, the mouth hooks.

Medial: Referring to, or at the middle of a structure.

Medio-dorsal: Relating to the median plane and the dorsal plane.

Membranous: Tissue which is thin, pliable and semi-transparent; like a membrane.

Meristem: Plant tissue characterized by frequent cell division, producing cells that become differentiated into specialized tissues.

Mesonotum: The upper surface of the second (middle) thoracic segment (mesothorax) of the insect body.

Mesophyll: The photosynthetic tissue of a leaf, located between the upper and lower epidermis. Mesophyll is commonly differentiated into palisade parenchyma and spongy parenchyma.

Mesothorax: The second or middle thoracic segment which bears the middle legs and anterior wings.

Metascutum: Posterior part of shield or shield-like structure.

Metathorax: The third (and last) segment of the thorax.

Microconidium (pl. microconidia): The smaller of two kinds of conidia formed by certain fungi.

Micropyle: A very small opening in the outer coat of an ovule, through which the pollen tube penetrates; the corresponding opening in the developed seed; one of the minute openings in the insect egg, through which spermatozoa enter in fertilization.

Microtrichium (pl. microtrichia): Small, sclerotized non-innervated cuticular projects on the body and wings of insects; also found on the tracheae.

Mildew: Thin coating of mycelial growth and spores on the surfaces of infected plant parts.

Mollicute: One of a group of prokaryotic organisms bounded by flexuous membranes and lacking cell walls (phytoplasmas and spiroplasmas).

Molt: A process of shedding the exoskeleton, ecdysis.

Monoclonal antibody: Antibody produced from clones of a single antibody-producing cell.

Monocotyledonous: An embryo having a single cotyledon.

Mosaic: Disease symptom characterized by non-uniform coloration, with intermingled normal, light green and yellowish patches, usually caused by a virus; often used interchangeably with mottle.

Mucro: Nematodes: A stiff or sharp point abruptly terminating an organ.

Multivoltine: Pertaining to organisms with many generations in a year or season.

Mutic: Without a point or pointed process; blunt.

Necrotic: Death of cells or tissue, usually accompanied by black or brown darkening.

Nectary: A nectar-secreting gland in a flower.

Neonate: Newly born individual.

Nematode: Non-segmented roundworm (animal), parasitic on plants or animals, or free living in soil or water.

Nerve ring: The center of the nervous system of nematodes that encircles the esophagus; composed largely of nerve fibers and associated ganglia.

Niche: 1) The physical and functional "address" of an organism within an ecosystem; or, where a living thing is found and what it does there; 2) the role an organism fills in an ecosystem.

Nucleolus (pl. nucleoli): A round or oval body in the nucleus of a eukaryotic cell; consists of DNA and RNA and produces ribosomal RNA.

Obclavate: Inversely clavate, widest at base.

Obovoid: Egg-shaped, with the narrow end outward.

Obligate: Restricted to a particular set of environmental conditions, without which an organism cannot survive. (e.g., an obligate parasite can survive only by parasitizing another organism.)

Obovate: A roughly elliptical shape with the terminal half broader than the basal.

Obpyriform: Inverse pear-shaped.

Ocellus (pl. ocelli): A simple eye of an insect or other arthropod.

Odontostylet: See stylet. Synonymous with onchiostylet.

Onchiostylet: Nematodes: A stylet developed from a special cell in the anterior part of the esophagus from which it moves into place during each molt.

Oocytes: Female germ cell.

Oogonium (pl. oogonia): Female gametangium of oomycetes, containing one or more gametes.

Oospore: Thick-walled, sexually-derived resting spore of oomycetes.

Ooze: Mass of bacterial cells mixed with host fluids.

Orbit: An imaginary border around the insect eye, the narrow part of the vertex adjacent to the margin of the compound eye.

Organelle: A membrane-bound structure within a cell having a specialized function, e.g. mitochondria and chloroplasts.

Ostium bursae: Ostium is the external genitalic opening of female Lepidoptera. Bursae the opening of the bursa copulatrix in Lepidoptera, equivalent to the vulva of female insects having the genital opening on the eighth segment.

Oviparous: Lay eggs.

Ovoid: Egg-like in shape or appearance.

Oviposit (oviposition): To deposit or lay eggs or ova. The act of depositing eggs.

Ovipositor: The external, tubular part of the female reproductive system through which eggs are passed. The ovipositor may be rigid and fixed in length or flexible and telescopic.

Ovisac: A receptacle for eggs.

Ovum: 1) A female gamete that corresponds to the male gamete; 2) the cell produced in the female reproductive system and which is capable of developing first into an embryo and ultimately into another individual of the same kind.

Palea: The small upper bract enclosing the flower of a grass.

Palisade mesophyll: A layer of columnar cells rich in chloroplasts found beneath the upper epidermis of foliage leaves.

Palp (pl. palpi): Finger-like, usually segmented appendage of the maxilla (maxillary palp) and labium (labial palp).

Papilla (pl. papillae): A hump or swelling.

Papillae anales: Lepidoptera: A pair of lobes at the apex of the female abdomen which are used in oviposition.

Paraphysis: An elongate sterile cell or hypha present in some fruiting bodies of fungi.

Parasite (adj. parasitic): Organism that lives in intimate association with another organism on which it depends for its nutrition; not necessarily a pathogen.

Parasitoid: A parasite that kills its host.

Parenchyma: The primary tissue of higher plants, composed of thin-walled cells and forming the greater part of leaves, roots, the pulp of fruit, and the pith of stems.

Parthenogenesis: Process of reproduction by the development of an unfertilized egg.

Parthenogenic: Pertaining to parthenogenesis.

Pathogroup: A pathogen group distinguished by host specificity.

Pathovar: A subdivision of a plant pathogenic bacterial species defined by host range; pathovar for bacteria is equivalent to *forma specialis* for fungi.

PCR (acronym for polymerase chain reaction): A technique used to amplify the number of copies of a specific region of DNA in order to produce enough of the DNA for use in various applications such as identification and cloning.

Peduncle: Stalk or main stem of an inflorescence; part of an inflorescence or a fructification.

Pericarp: The wall of a fruit, derived from the maturing ovary wall.

Pericycle: Layer or layers of cells between the phloem and the endodermis of roots, giving rise to branch roots.

Perithecium (pl. perithecia): Flask-shaped or subglobose, thin-walled fungus fruiting body (ascocarp) containing asci and ascospores; spores are expelled or released through a pore (ostiole) at the apex.

Peritreme: 1) The margin of a shell opening; 2) the cuticular margin which surrounds a spiracle.

Pestiferous: Producing or breeding infectious disease. Infected with or contaminated by an epidemic disease.

Petiole: 1) Botany: stalk portion of a leaf; 2) Insect: Apocrital Hymenoptera; the narrow second (and sometimes third abdominal segments that precede the gaster) forming the 'waist'.

Phagocyte: A cell that is able to ingest and destroy foreign matter, including bacteria.

Phagocytic: Capable of functioning as a phagocyte.

Phasmid: Nematodes: A pore-like structure located in the lateral field of the posterior region of nematodes belonging to the class Secernentea. Function is believed to be sensory. Sometimes called precaudal glands.

Pheromone: A substance given off by one individual that causes a specific reaction by other individuals of the same species, such as sex attractants, alarm substances etc.

Phloem: The vascular tissue in vascular plants, that conducts and distributes sugars and other dissolved foods from the places the food is produced to the places the food is needed or stored.

Phytoalexin: A low molecular weight, antimicrobial compound synthesized by and accumulating in higher plants exposed to certain microorganisms (pathogenic and nonpathogenic).

Phytophagous: Plant eating.

Phytotoxic: Poisonous to plants.

Placenta: Site of attachment of the ovule to the ovary wall. Plasma membrane; the outer limiting membrane of a cell. The tissue in the female reproductive organ of a plant that produces the ovules.

Plica (pl. Plicae): A fold, or convolution or wrinkle in a structure or surface.

Plumule: The primary bud of an embryo or germinating seed.

Polar: At one end or pole of the cell.

Polyclonal antibody: A preparation containing antibodies against more than one epitope of an antigen.

Polyphagous (Polyphagy): Eating many kinds of food.

Polyphenols: Group of vegetable chemical substances, characterized by the presence of more than one phenol group. The reactions of these molecules help to produce gelatines, alkaloids and other proteins.

Polyvoltine: See multivoltine.

Postocellar area: Hymenoptera: the region on the dorsal aspect of the head bounded by the ocellar furrow, vertical furrows and the caudal margin of the head.

Postocular: Pertaining to the structure or color posterior of the compound eyes.

Posterior: A term of position pertaining to a structure situated behind the axis. Toward the rear, caudal or anal end of the insect; opposed to anterior.

Potyvirus: (Siglum of potato virus Y). Member of a large group of plant viruses with flexuous particles containing a single molecule of linear RNA, most of which are transmitted by aphids in a noncirculative manner.

Process (pl. processus): A projection from the surface, margin or appendage.

Pronotum: The upper (dorsal) plate of the prothorax.

Prepupa (pl. prepupae): 1) A quiescent instar between the end of the larval period and the pupal period; 2) an active but non-feeding stage in the larva of the Holometabola; 3) a full-fed larva.

Proleg: 1) Any process or appendage that serves the purpose of a leg; 2) specifically, the pliant, non-segmental abdominal legs of caterpillars and some sawfly larvae. Not true segmented appendages.

Prothorax: The first segment of the thorax.

Prothoracic plate: Dorsal part of 1st thoracic segment of, for example, lepidopterous larva with cuticle thickened, often coloured, and shield-shaped.

Protonymph: The second instar of a mite.

Protoplasm: Living contents of a cell.

Protoplast: Living cell exclusive of a wall.

Pseudoparenchyma: A mass of hyphae arranged together to form a tissue like structure. A dense tissue formed by hyphae becoming twisted and fixed together where the hyphal components of the tissue are no longer distinguishable.

Ptilinum: An inflatable organ capable of being thrust out through the frontal suture just above the root of antennae, at emergence from the pupae.

Pulverulent: Appearing as though covered with a fine powder.

Pulvinate: Cushion, cushion-shape, flattened pads or pad-like.

Pupa (pl. pupae): The stage between the larva and adult in insects with complete metamorphosis, a nonfeeding and usually inactive stage.

Puparium: The thickened, hardened barrel-like larval skin within which the pupae is formed.

Pupation: Becoming a pupa.

Pustule: A blister-like spore mass breaking through a plant epidermis.

Pygopod: Any appendage of the tenth abdominal segment.

Pycnium (pl. pycnia): Globose or flask-shaped haploid fruiting body of rust fungi bearing receptive hyphae and pycniospores.

Raster: Scarabaeoid larvae: a complex of specifically arranged bare areas, setae and spines on ventral surface of last abdominal segment, anterior of anus.

Raceme: A type of inflorescence in which flowers are formed on individual stalks along a main axis or peduncle.

Radicle: Part of the plant embryo that develops into the primary root.

Reniform: Kidney-shaped.

Restriction fragment length polymorphism (RFLP): A variation in DNA sequence that is easily recognized because it occurs at a site where a restriction enzyme cuts a specific sequence, producing DNA fragments of varying lengths. RFLP's often serve as genetic markers.

Reticulate: Descriptive of surface sculpture, usually the insect’s integument, that is covered with net-like lines.

Rhinarium (pl. rhinaria): 1) A nostril pieced or portion of the Nasus; 2) Odonata: lower portion of the clypeus; 3) Hymenoptera: elevated, keel-like ridges on the flagellar segments of the parasitic Hymenoptera antennae.

Race: Subgroup or biotype within a species or variety, distinguished from other races by virulence, symptom expression, or host range, but not by morphology.

Rachis: Elongated main axis of an inflorescence.

Rostral: Pertaining to a rostrum.

Rostrum: Beak or snout such as weevils.

Rugose: Wrinkled, roughened.

Rust: A disease caused by a specialized group of basidiomycetes that often produces spores of a rusty color.

Saprophyte (adj. saprophytic): Organism that obtains nourishment from non-living organic matter.

Scab: Roughened, crustlike diseased area on the surface of a plant organ.

Scarification: The physical or chemical treatment given to some seeds in order to weaken the seed coat sufficiently for germination to occur.

Sclerenchyma (adj. sclerenchymatous): Tissue made up of thick-walled plant cells.

Sclerite: A hardened body wall plate bounded by sutures or membranous areas.

Sclerotized: Hardened.

Scutellum: A sclerite of the thoracic notum; the mesoscutellum appearing as a more or less triangular sclerite behind the pronotum, especially in Hemiptera.

Semi-looper: A caterpillar in which 1-2 pairs of the abdominal legs are absent and movement is restricted to progression only in small loops (of the Noctuoidea superfamily).

Semilunar: In the form of a half crescent.

Senesce: To decline in stature, vigor and capacity following maturity.

Sensillum: 1) A simple sense organ or sensory receptor found on various appendages and tagmata of the insect body. 2) a complex, bilaterally symmetrical sexually-dimorphic structure found on the tenth tergum of Siphonaptera. Covered with short, tapering spines (termed microtrichia), circular pits which give rise to longer setae (termed trichobothria) and a dome-like cupola. Functions as a compound sense organ.

Septate: With cross walls; having septa.

Septum (pl. septa): Dividing wall; in fungi, cross wall.

Serpentine mines: Narrow, winding leaf mines which increase in width with larval growth.

Serotype: A subdivision of virus strains distinguished by protein or a protein component that determines its antigenic specificity.

Sessile: Used in reference to a leaf, leaflet, flower, floret, fruit, ascocarp, basidiocarp, etc., without a stalk, petiole, pedicel, stipe or stem; (of nematodes) permanently attached; not capable of moving about.

Seta (pl. setae): A bristle; commonly known as hairs.

Sheath: The extension of the leaf that surrounds the stem.

Single stranded, positive sense RNA: Also known as a sense-strand RNA virus, a virus whose genetic information consists of a single strand of RNA that is the positive (or sense) strand which encodes mRNA (messenger RNA) and protein. Replication in positive-strand RNA viruses is via a negative-strand intermediate. Examples of positive-strand RNA viruses include polio virus, Coxsackie virus, and echovirus.

Siphunculus (pl. siphunculi): The instrument of suction in sucking insects.

Somatic: Referring to vegetative or non-sexual stages of a life-cycle.

Spathe: An enlarged leaf-like bract that surrounds or partially encloses an inflorescence.

Spatulate: Spatula-shaped.

Spermatheca: A sac, duct or reservoir within the female that receives spermatozoa during copulation. Variable in number and morphology and capable of storing spermatozoa for periods up to several years.

Spinneret: Opening of the silk gland, found on the caterpillar's lower lip. It's used to create the silk pad to which the chrysalis attaches.

Spicule: Male copulatory organ. Sometimes called speculum.

Spiciseta (pl. spicisetae): One of three types of body setae found on larval Dermestidae. Variable in length and sometimes exceeding the body length; commonly sharply pointed, overlapping scales.

Spiracles: A breathing pore; in the plural the lateral openings on the segments of the insect body through which air enters the trachaea.

Spongy parenchyma: Cynipid galls: the material occupying the central portion of a gall and constituting the major material of all the spongy and more hollow oak-apples of the genus *Cynips*.

Spore: A specialized reproductive body in fungi (and some other organisms), containing one or more cells, capable of developing into an adult.

Sporulate (sporulation): To produce spores.

Squama (pl. squamae): 1) A scale or scale-like structure; 2) a broad, flat sclerite attached to an organ, appendage or structure.

Stele: Central cylinder of vascular tissue (especially in roots).

Stigma: Portion of a flower that receives pollen and on which the pollen germinates.

Stipe: Stalk.

Stipule: One pair of leaf-like structures, spines, glands, or scales at the leaf base or along a petiole.

Stolon: A slender, horizontal stem that grows close to the soil surface; in fungi, a hypha that grows horizontally along the surface.

Stria (pl. striae): Descriptive of the surface sculpture, usually the insect's integument, that is marked with numerous parallel, fine, impressed lines.

Strigula: A fine, short, transverse mark or line.

Stroma: Compact mass of mycelium (with or without host tissue) that supports fruiting bodies or in which fruiting bodies are embedded.

Stunting: Reduction in height of a vertical axis resulting from a progressive reduction in the length of successive internodes or a decrease in their number.

Subepidermal: Beneath the epidermis.

Supra-anal: Located above the anus.

Suture: Gastropods, the spiral line that marks the junction of the whorls; in chitons, the junction between girdle and valves.

Sympodial: Pertaining to proliferation of axes, in which each successive spore or branch develops behind and to one side of the previous apex where growth has ceased.

Syncytium (pl. syncytia): A multinucleate structure in root tissue formed by dissolution of common cell walls induced by secretions of certain sedentary plant-parasitic nematodes, e.g. cyst nematodes.

Tail: Nematodes: the portion of the body between the anus and the posterior terminus.

Tarsus (pl. tarsi): The leg segment immediately beyond the tibia, consisting of one or more segments or subdivisions.

Tergite: A dorsal sclerite or part of a segment, especially when such part consists of a single sclerite.

Tegumen: Lepidoptera: the tergum in male genitalia. A structure shaped as a hood or inverted trough, positioned dorsad of the anus; the uncus articulates with its caudal margin, derived from the ninth abdominal tergum.

Teleomorph: The sexual form in the life cycle of a fungus.

Teneral: Describing the imago or adult shortly after emergence from the nymphal or pupal stage when the integument is not hardened or its color has not matured.

Termen: The outer margin of a wing, between the apex and the posterior or anal angle.

Testa: Seed coat.

Testaceous: A dull brick red or brownish red color.

Testis: A male reproductive organ in which spermatozoa are produced.

Thorax: The body region behind the head, which bears the legs and wings.

Tibia (pl. tibiae): The fourth segment of the leg, between the femur and tarsus.

Tibial spur: The spur or spurs frequently borne near to or at the distal end of the tibia.

Tiller: A lateral shoot, culm, or stalk arising from a crown bud; common in grasses.

Tornus: Lepidoptera: the junction of the tegmen and the dorsum of the wing, hind or anal angle.

Toroid (toroidal): A doughnut shaped coil.

Transverse: Pertaining to structures which are wider than long; running across or cutting the longitudinal axis at right angles.

Truncate: Pertaining to structures which end abruptly as if cut at a right angle to the longitudinal axis.

Tubercle: A little solid pimple or small button, in Sphecoidea rounded lobes of the dorsal lateral margin of the pronotum; in caterpillars, body structures of the character, sometimes bear setae.

Umbilicus: Hole about which the shell spirals

Unilocular: With one cavity.

Uninucleate: Having one nucleus.

Urediniospore (also urediospore, uredospore): The asexual, dikaryotic, often rusty-colored spore of a rust fungus, produced in a structure called a uredinium; the "repeating stage" of a heteroecious rust fungus, i.e. capable of infecting the host plant on which it is produced.

Uredinium (also uredium; pl. uredinia): Fruiting body (sorus) of rust fungi that produces urediniospores.

Urogomphi: Fixed or mobile processes found on the terminal segments of certain larvae; variously termined styli, cerci, pseudocerci, corniculi.

Valva: Harpagones or two lateral sclerites which cover the ovipositor when not in use.

Vascular bundle: Strand of conductive tissue, usually composed of xylem and phloem (in leaves, small bundles are called veins).

Vector: Literally a bearer; specifically a host of a disease transmissible to another species of organism.

Vein clearing: Disappearance of green color in or around leaf veins (a common symptom associated with virus infection).

Ventral: Pertaining to the under surface of abdomen.

Vermiform: Worm-shaped.

Vesica: Lepidoptera: the penis, or terminal part of the aedeagus. Vesica is membranous and eversible; typically held within the tubular part of the aedeagus but everted and inflated during copulation.

Virion: Complete virus particle.

Virus: A submicroscopic,intracellular, obligate parasite consisting of a core of infectious nucleic acid (either RNA or DNA) usually surrounded by a protein coat.

Viviparous: Pertaining to organisms which bear living young; opposed to organisms which lay eggs.

Vomitoxin: A toxic substance produced by mold.

Zoospore: Fungal spore with flagella, capable of locomotion in water.

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Appendix A: Diagnostic Resource Contacts

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